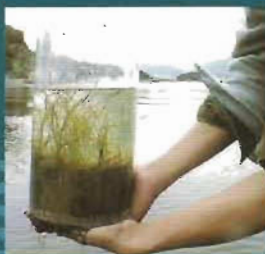


A NaGISA Handbook

Sampling Biodiversity in Coastal Communities

NaGISA Protocols for
Seagrass and Macroalgal Habitats

Edited by
P. R. Rigby, K. Iken
and Y. Shirayama



Kyoto University Press



RIDGE BOOKS

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Cover photo 1: Sampling a rocky intertidal site on Kodiak Island, USA
(*photo courtesy of D. Kubiak 2004*)

Cover photo 2: A seagrass core from Tanabe Bay, Japan (*photo courtesy of P.R. Rigby 2003*)

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Table of Contents

<i>List of Figures</i>	vii
<i>Foreword</i>	ix
Part I Introduction	
Section 1 Introduction to the Text	1
Section 2 The NaGISA Concept	5
Section 3 Biodiversity and Sampling Design	7
Part II Rocky Shore and Seagrass Bed Ecology and Sampling Protocols	
Section 4 Rocky Shore Ecology	13
Section 5 NaGISA Rocky Shore Protocol	17
Section 6 Seagrass Bed Ecology	22
Section 7 NaGISA Seagrass Protocol	25
Part III Specific Details for Working with the Primary Target Groups	
Section 8 Macroalgae	33
Section 9 Seagrasses	37
Section 10 Echinodermata	42
Section 11 Polychaeta	47
Section 12 Cnidaria	52
Section 13 Mollusca	56
Section 14 Decapoda	59
Part IV Specific Details for Working with the Secondary Target Groups	
Section 15 Porifera	69
Section 16 Amphipoda	73
Section 17 Isopoda	76
Section 18 Meiobenthos	80
Part V Beyond NaGISA; Further Information and Optional Protocols	
Section 19 Practical Coastal Physical Oceanography for Ecologists	85
Section 20 Guide to Identification and Surveying of Nearshore Fishes	90
Section 21 Sampling and Monitoring Rhodolith Beds	93

Part VI Reference Tools

Section 22	Priorities within the Protocols	101
Section 23	NaGISA Field Data Sheet	103
Section 24	Glossary of Methods	104
Section 25	Glossary of Terms	108
Section 26	Bibliography	117
Section 27	Online References	130
Section 28	Contributors	138
Section 29	Index	144

List of Figures

Fig. 5.1	Rocky Shore Layout	18
Fig. 5.2	Rocky Shore Quadrat Set	19
Fig. 5.3	Macroalgae: Activity Flow Chart	20
Fig. 6.1	Model of Seagrass Genera Arranged by Size	23
Fig. 7.1	Seagrass Bed Transect Layout	26
Fig. 7.2	Seagrass Bed Quadrat Set	27
Fig. 7.3	Seagrass Bed Cores	27
Fig. 7.4	Sampling Horizontally Distributed Seagrass Beds	28
Fig. 7.5	Seagrass Bed: Activity Flow Chart	29
Fig. 8.1	General Morphology of Macroalgae	34
Fig. 9.1	Key Morphological Characteristics of Strap-like Leaf Blade Seagrasses	38
Fig. 9.2	Key Characteristics of Oval Shaped Leaf Blade Seagrasses	39
Fig. 10.1	Representative Echinoderms	44
Fig. 11.1	Phyllococidae, Syllidae, Polynoidae, & Terebellidae	48
Fig. 11.2	Polychaete Dissection	49
Fig. 12.1	A Medusa of a Hydrozoan, a Sea Anemone & Hydroid Colonies	53
Fig. 13.1	Parts of the Bivalve Shell	57
Fig. 13.2	Parts of the Gastropod Shell	58
Fig. 13.3	Parts of the Polyplacophoran Shell	58
Fig. 14.1	General Body Plan of Dendrobranchiata	60
Fig. 14.2	General Body Plan of Caridea	61
Fig. 14.3	General Body Plan of Stenopodidea	62
Fig. 14.4	General Body Plan of Brachyura	63
Fig. 14.5	General Body Plan of Anomura	64
Fig. 14.6	General Body Plan of Palinura	65
Fig. 14.7	General Body Plan of Thalassinidea	65
Fig. 15.1	The Sponge Body Plan	70
Fig. 16.1	General Amphipod Body Plan	74
Fig. 17.1	Cirolanid Isopod	77
Fig. 17.2	Arcturid Isopod	77
Fig. 18.1	Representative Species of Meiofaunal Phyla	81
Fig. 20.1	Kelp Bass	91
Fig. 23.1	NaGISA Field Data Sheet	103

Foreword

Natural Geography In Shore Areas (NaGISA) was one of the first projects to participate in the Census of Marine Life (CoML) (www.coml.org). With the goal to discover *what was, what is, and what will be* living in the world's oceans, CoML has turned to NaGISA to help *discover the world's nearshore*. NaGISA's simple, standardized protocols and focus on widespread nearshore habitats make it perfectly suited to assess nearshore biodiversity on a global scale. It also made it the ideal choice of ambassador for CoML. NaGISA is a project that could be conducted by anyone anywhere and thus brings global grassroots participation to CoML. While CoML has expanded to include over 18 projects, ranging from the nearshore to the continental margins and abyssal plains, and targets over a billion cubic kilometers of under-sampled mid-ocean waters, NaGISA still remains the most hands-on and international of all the projects.

Although the technology for sampling in the nearshore is straightforward, the technology for analyzing the samples can be quite advanced. NaGISA faces the challenge of accumulating and preserving traditional taxonomic expertise for a large number of different taxa. However, NaGISA's ongoing work on advanced imaging technologies and its future cooperation with other census projects such as the marine microbe project and the Barcode of Life will be the key to rapid reassessment of biodiversity change. NaGISA's current work will lay the foundation for combining these advances with traditional taxonomy and will allow monitoring of both the loss of diversity and the appearance of invasive species in a matter of days instead of months. NaGISA participation in the Census also means that the information, whether data or highly resolved images will be publicly accessible through the Ocean Biogeographic Information System (www.iobis.org), the data portal for all CoML projects.

It is easy to get excited about the unknown, and the mysteries that lie within our reach on the ocean's doorstep are no exception. There are millions of kilometers of shoreline between the Arctic and the Antarctic that are just waiting to be sampled. The more sampling done the more surprises there will be: species that are found out of their previously documented ranges, unheard of relationships between species and maybe even species that have never been seen before.

I encourage all readers to enjoy their journey into the near unknown.

R.K. O'DOR, DALHOUSIE UNIVERSITY, CANADA

Part I

Introduction

Section 1 Introduction to the Text

Welcome to the nearshore as seen through the eyes of NaGISA. Whether you are planning to sample the coast for the first time or are a long-time NaGISA participant reviewing techniques before conducting an annual survey, we trust this text will prove useful. You will find the basic steps and procedures used to conduct standardized biodiversity studies in rocky shore and seagrass coastal communities along with introductory chapters on biodiversity, macroalgae and seagrass ecology. Written by premier coastal ecologists and taxonomists, this book has been organized as a text for field courses, a manual for coastal managers, and a reference guide for researchers studying biodiversity, attempting to inventory species or monitor their own local areas.

Chapters on the concepts of biodiversity, benthic ecology and data management complement the descriptions of habitat-based protocols that have been developed by researchers over the past ten years. The NaGISA protocols given here represent the minimum standard by which biodiversity should be monitored on a global scale and are written with flow charts and diagrams in order to ensure their replication. Following the standardized protocols are guides on how to prepare and identify specific taxonomic groups. These groups have been selected for special attention, in some cases because they are fairly simple to do, in others because they have never been done, but most often because their study will reveal fundamental aspects of the benthic ecosystem. The guides have been written with the intent of instructing life science students and researchers on handling specimens outside of their own field of expertise. The introductory tone is continued throughout the text, and is supported by the inclusion of a glossary of methods and compilation of selected references, field checklists and example data sheets, included to ease even the newest researcher's transition into the field.

The global study of benthic communities requires international cooperation and the proliferation of standardized techniques. Although we initially selected two globally occurring habitats as our standard we have been inundated with requests to authenticate protocols for a variety of coastal habitats. We include some of these as supplementary protocols here as they complement our overall goals including the elucidation of a baseline of global biodiversity. We hope they will be of assistance to those who wish to go over and above the global minimum.

All suggestions or comments or requests are most welcome and should be directed to our Headquarters:

NaGISA HQ
Kyoto University
459 Shirahama
Nishimuro, Wakayama
649 2211 Japan

P.R. RIGBY, KYOTO UNIVERSITY, JAPAN

Section 2 The NaGISA Concept

Biodiversity and global strategies for ensuring its stability have been discussed in many forums and on many levels. Loss of diversity has become a political rallying point, recognized on the world stage when the Convention of Biological Diversity (CBD) was established. This global call to establish national species inventories and data collection has inspired national initiatives by numerous individual signatory members but as yet lacks a system to ensure its global implementation or an international body to disseminate findings.

In 2001, the Western Pacific (WestPac) division of DIVERSITAS, an organization dedicated to the preservation of global biodiversity, inaugurated the International Biodiversity Observation Year (IBOY). The most prominent outcome of IBOY's activities was the recognition of an immediate need to conduct a standardized assessment of global diversity. The coastal zone was one of three selected target habitats and outlines of the protocols needed were drawn up by a group of designated coastal scientists (Shirayama *et al.* 2002). In 2002, when the Census of Marine Life (CoML) was looking for its first suite of projects, the ideas laid out during IBOY were adopted and the NaGISA project was formed.

To facilitate the rapid growth of NaGISA a structural hierarchy was introduced. There are currently three levels in the system: International, Regional and Local. NaGISA Headquarters manages the overall concept and direction of the global NaGISA project and is situated at the Seto Marine Biological Laboratory at Kyoto University, Japan. The regional level is divided into eight centers: Western Pacific, Eastern Pacific, European Seas, South American Seas, Caribbean Sea, Indian Ocean and Atlantic Ocean. These regional centers coordinate NaGISA activities in the various regions and act as the bridge between HQ, national groups, local areas and sites. The local level is where most of the activity happens as students, volunteers,

scientists and local support groups are drawn into NaGISA activities: sampling and sample analysis and marine education and awareness programs.

The specific strength of NaGISA is the adoption of standardized sampling and sample-processing protocols to be used by local members integrated in a global network. This standardization will allow biodiversity to be compared within local areas and between regions and will for the first time elucidate geographic patterns of biodiversity along latitudinal and longitudinal directions at scales ranging between a few meters and thousands of kilometers. The data collection will be ongoing and will serve as a living archive of global coastal marine biodiversity information from which students, researchers and stakeholders can receive continuously updated information on local, regional and global biodiversity and its change over time. The hope is that NaGISA will grow from a network of individually funded scientific endeavors to a governmentally supported network, for which national budgets contribute to the study and understanding of national resources.

Y. SHIRAYAMA & P.R. RIGBY, KYOTO UNIVERSITY, JAPAN

Section 3 Biodiversity and Sampling Design

Biodiversity, short for biological diversity, is a common term used in various ways by ecologists, managers and the general public. In popular usage, biodiversity simply refers to the number of species that occur in a given area. When scaled-up to include the whole planet, biodiversity refers to all life on earth. This is an engaging term for a wide audience, including non-scientists, because it focuses attention on the importance of preserving life on earth and appeals to us to take action to counteract species extinction.

Ecologists usually refer to biodiversity as the variability that occurs at different levels of biological organization, including the processes that maintain this variability (Gaston 1996). Thus, biodiversity is the number of genes, organisms, populations, species and habitats that occur in a given seascape or landscape, including the interactions between each of these levels of biological organization and their environment over a hierarchy of spatial and temporal scales. This is summarized by the definition of biodiversity given by the United Nation Environment Programme (UNEP) [Heywood 1995; see Magurran (2004) for a detailed history of the concept]:

“Biological diversity” means the variability among living organisms from all sources including, inter alia, terrestrial, marine and other aquatic systems and the ecological complexes of which they are part; this includes diversity within species, between species and of ecosystems.

Why is biodiversity considered so important? Intuitively, the more diversified life is, the greater the ability of natural systems to deliver goods and services. These include, among others, food and medical resources, drinkable water, clean air and fertile soils. Biodiversity also contributes to the productivity of natural and agricultural systems in various ways, such as through the effects of pollinators or the control of pests by natural predators. In addition

it has an intrinsic aesthetic value. Loss of biodiversity is therefore of considerable concern to ecologists, policy-makers and the general public because it threatens the functioning of the current system. The current rate at which species go extinct seems unprecedented and several of these extinctions are suggested to be linked to human activities (Chapin *et al.* 1998). Destruction of natural habitats, loss of strong interacting species (such as habitat formers and keystone species), introduction of non-native species, over-exploitation of natural resources (such as over-fishing), urbanization, pollution and climate change are all potential threats to biodiversity.

Ecologists have developed specific research programmes to understand the causes and to predict the consequences of loss of biodiversity, with the ultimate goal of providing the basic scientific understanding necessary to underpin better policies of management and conservation of natural and agricultural resources. These programs have framed the relationship between biodiversity and the functioning of natural systems in a scientifically tractable way and have used a more synthetic definition of biodiversity. This has commonly been redefined as the numbers, identities and relative abundances of species (or higher taxa combining morphologically or functionally similar species) in a habitat. The scientific hypothesis under scrutiny is that biodiversity is positively related to measurable properties of natural systems like productivity, nutrient cycling, stability and resilience (defined as the ability of a system to recover from disturbance). Although a detailed discussion of the outcomes of these studies is beyond the scope of this introduction, this hypothesis is supported by some, but not all the investigations that have been conducted in aquatic and terrestrial environments. The interested reader may refer to literature on this topic (Schläpfer and Schmid 1999; Waide *et al.* 1999; Loreau *et al.* 2002; Worm and Duffy 2003). Of more interest to us is the methodological rationale of the sampling design on which these studies are based.

Observational studies examining biodiversity-ecosystem functioning relationships (BEF) in natural systems face two major methodological challenges: one is the estimation of biodiversity (usually the number of species) in a given area; the other is the comparison of these estimates over space and/or time. Since it is impossible to enumerate systematically the number of all the species (or higher taxa) that are present in a given habitat, which would require the collection of all the theoretically possible observations that could be conducted in that habitat, the assessment of biodiversity relies on sampling. Thus, estimates of total biodiversity must be extrapolated from sample data. Various techniques are available for this, including species-area accumulation curves, rarefaction methods, moment-based and non-parametric estimators [the interested reader may refer to Colwell *et al.* (2004) and to Magurran (2004) for further details]. These techniques share the same basic methodology in the way the data are collected. The data are drawn from a collection of samples distributed in space and/or time according to specific criteria determined by the particular specific hypothesis of interest. Whilst the techniques for collecting samples differ depending on the system being investigated (for example, quadrates on rocky shores versus cores in soft sediment), the principles of sampling design do not. Because these principles establish logical linkages between the spatial (or temporal) arrangement of sampling units and the specific hypothesis of interest, they are at the core of any comparative study of biodiversity and need to be clearly understood before embarking in this type of analysis.

A common approach to the study of BEF is that of comparing patterns of biodiversity across latitudinal gradients or other natural gradients like salinity, exposure to waves or nutrients. The logic underpinning this approach is that there is an *a priori* expectation that biodiversity and associated functional properties change along gradients. For example, some evidence indicates that tropical systems have more species than those occurring at higher latitudes, and if there is a positive correlation between biodiversity and productivity on large spatial scales, then the biodiversity-productivity relationship can be expected to follow the same pattern. To test this hypothesis it is necessary to sample a number of localities at different latitudes and to measure productivity and diversity in a number of replicate samples in each locality. For example if five replicate samples per tidal height are collected at sites established along a latitudinal gradient, then latitude is the factor that is expected to account for a significant amount of the variability in the data. Such a factor consists of a number of levels (the various sites) representing groups of observations (the replicates) exposed to the same condition with respect to the source of variability that the factor represents (latitude). Because the levels of the factor 'Latitude' must be arranged along a latitudinal gradient to test the hypothesis of interest, 'Latitude' is a fixed factor. In other words, the sites create a latitudinal gradient and each site differs from the others because of its unique property of representing a specific latitude.

Of course, sites may also differ from each other for many reasons that may have nothing to do with latitude (for example, oceanography, geological history, etc.). For this reason, a genuine latitudinal gradient cannot be represented by a single site at each latitude because this would confound latitudinal effects with the intrinsic differences among sites due to other factors. This is a very important point that is often neglected in large-scale studies on biodiversity. Technically, the problem is known as pseudo-replication, following the terminology introduced by the seminal (and recommended) paper by Hurlbert (1984). Site is the appropriate level of replication to test the hypothesis of interest, not samples within a site. If there is only one site for each latitude, the sampling design is pseudo-replicated. The appropriate design must therefore include replicate sites within each latitude. NaGISA protocols demand that at least three replicate sites that represent the habitat of interest should be chosen randomly within each latitude. 'Site' now becomes a second factor in the design with three levels (three sites within each latitude). Note that there is no particular interest in any of these sites, they are included in the study with the purpose of estimating natural variability within latitudes. Any set of sites would take care of this task. Including 'Site' in the design makes it possible to disentangle the variation due to the latitudinal gradient from other sources of variability that are quantified by the differences among sites. Two things must be noted in this design. First, 'Site' is not a fixed factor because its levels are selected randomly from a population of possible levels. It is a random factor. Second, each site can occur at just one latitude (by definition, a site cannot span several latitudes); 'Site' is nested within 'Latitude'. In summary, this sampling design consists of a fixed nesting factor (Latitude, with as many levels as latitudes sampled), a random nested factor ('Site', with three levels) and five replicate samples per tidal height in each locality. Samples are also a random source of variation since they are used to quantify the variability in each site.

The nested sampling design just illustrated can be extended to include several spatial, temporal, or combined spatial and temporal scales. This is relevant to NaGISA where patterns of biodiversity are examined both at global and local scales on rocky shores and in seagrasses. The objective is to estimate variability at each scale and to identify the scale(s) at which most of the variability occurs. Because different ecological processes generate variability at different scales, identifying the relevant scales of variability in patterns of biodiversity or in BEF focuses attention on the potentially important causal processes. This provides important clues for directing subsequent experimental research.

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Part II

Rocky Shore and Seagrass Bed Ecology and Sampling Protocols

Section 4 Rocky Shore Ecology

INTRODUCTION

Shorelines are areas of transition between the ocean and land. Rocky areas dominate much of the world's nearshore. The communities that live in these areas are known for their high biodiversity, as the rocks provide the hard substrate necessary for the attachment of sessile organisms, particularly macroalgae. These nearshore macroalgal communities are very productive. An example of this productivity is the primary production in the giant kelp (*Macrocystis pyrifera*) forests in California, which ranges between 250 to 1500 g C/m²/yr (Mann 1982). Macroalgal communities are important habitat for many invertebrates, fishes and marine mammals. These organisms use macroalgae for food and shelter. Many macroalgal habitats are known to be nursery grounds for recreational, commercial and subsistence invertebrates and fishes. The nearshore can be said to have two major areas: intertidal and subtidal. The intertidal zone is alternately exposed to air and water while the subtidal zone is continuously submerged.

The intertidal area ranges from the splash zone to mean low water level. The spatial extent of this area varies depending on its slope and tidal exchange. Due to variations of slope (steeper slopes have less aerial intertidal space) and tidal exchange (which can range from a few centimeters to as large as 10m) the spatial extent of the intertidal zone can vary greatly.

The intertidal area can be divided into four zones: splash, high, mid and low.

The *splash zone* is the area that is reached by the ocean's salt spray. Only the highest tides and storm waves contact this zone. Most of this zone is bare rock but there are some common organisms found here, including a few species of periwinkles, limpets, and barnacles. Some species of lichens and one green alga (*Prasiola* spp.) also are common in this zone.

The high-tide zone generally provides a home to a greater variety of life than the splash zone. This zone is exposed to waves and is covered with water only during (most) high tides. Only organisms that can withstand rough waves and long exposure to air can survive. The most common animal in this zone is the barnacle, which cements itself to the substrate and closes its test during low tides to avoid desiccation. The algal community is not diverse in this zone and usually consists of ephemeral species such as the green algae in the Order Ulvales and brown algae of the Order Fucales. These leafy or thickly branched algae provide protection from desiccation to resident invertebrates by keeping the animals damp and shaded.

The mid-tide zone is generally more diverse than the higher zones because it is covered and uncovered by the tides on a shorter, more regular cycle. Organisms living here have adapted well to this change in emersion. Common animals in this zone can include many species of anemones and mollusks (including chitons, limpets, mussels, and gastropods). During low tide, anemones can conserve water by closing up and covering themselves with small rocks and shell debris. Molluscs can protect themselves by using their powerful foot to secure themselves close to the substrate so as to protect their soft parts or can move to moist areas such as cracks or under macroalgae. Other molluscs have an operculum so that they can use to close their shells to avoid water loss. The most common macroalgae in the mid-tide zone are the Fucales or rockweeds, although others, particularly many red algae, exist depending on the region.

The low-tide zone is the most diverse of the intertidal zones. It is usually submerged and only exposed to air during unusually low tides. Because of the longer submersion period, organisms in this zone are not as adapted to intertidal living. Common animals found here can include seastars, urchins, crabs, gastropods, anemones, small crustaceans, and small fishes. Often these animals will migrate to the subtidal area during particularly low tides.

The subtidal area extends from below the low intertidal into and including the depths that support large macroalgae given the proper substrate (often as deep as 20m or more). Because the subtidal zone is continuously submerged, many subhabitats exist along both vertical (through the water column) and horizontal (along the substrate) depth gradients.

A *vertical depth gradient* can be understood by envisioning a cross-section of the subtidal area from the surface to the substrate. On the surface of a kelp forest, many birds and marine mammals are associated with the canopy forming blades of the large kelps. These animals forage in the midwater and at the bottom. Commonly associated with the midwater are phytoplankton, zooplankton and fish. Many fish utilize the structure afforded by the canopy and midwater kelps for protection and food. Connected with the kelp structure floating on the surface and in the midwater, many encrusting and mobile organisms use the blades and stipes for substrate and food. Encrusting bryozoans, mobile gastropods and limpets, and isopods are often abundant on mature kelp. The fauna associated with kelp holdfasts is incredibly diverse. Upward of 351 species from 213 families and 15 phyla were found in eighty *Ecklonia radiata* holdfasts in New Zealand (Anderson *et al.* in press). The most common groups were the mobile arthropods, annelids, mollusks and echinoderms. Similar species-rich communities have been reported from holdfasts of *Laminaria hyperborea*, *Durvillaea antarctica*,

and *Macrocystis pyrifera*. Similar to kelp holdfasts, upright coralline algae and other turfing algae that are attached to the substrate can contain high biodiversity (Dearn 1987).

A *horizontal depth gradient* is seen in the transition from the shallow subtidal to greater depths, even though the areas often have very patchy species distributions. This small-scale patchiness can often result in a community structure that has very high natural variation (Edwards 2004). In general though, shallower areas often have higher biodiversity than deeper regions because of the continual disturbance from surge and wave action. In these shallow areas, red and brown algae are abundant, as are seastars, urchins, mollusks and crustaceans. As depth is increased, storm surge and similar disturbance is decreased, however, other factors become more important. For example, as depth increases, light decreases. This change in light attenuation will cause associated changes in the algal community, which will have cascading effects on faunal community structure. Typically for the macroalgae, brown algae will become less abundant and certain species of red algae will prevail, particularly the corallines. Changes in algal community structure will affect faunal distribution and abundance.

FACTORS INFLUENCING COMMUNITY STRUCTURE

Biotic and abiotic factors influence the community structure of nearshore rocky communities, including algal and faunal composition and abundance. Below is a limited, but not all-inclusive discussion of some important controlling factors.

Biotic factors typically include predation (including grazing) and competition. Predation is typically viewed as a “top-down” control. Animals and plants are usually the prey of animals from the same zone or the one directly beneath it. Some of these predators can play keystone roles on the community by having extreme impacts although the predators’ densities are not very high. For example, studies have shown that seastars from low and subtidal zones can play a keystone role in determining the structure of the midtidal zone by preying on mussels that reside here (Paine 1974). Similarly, animals that can be considered top predators can have cascading effects throughout the community. For example, it has been hypothesized that killer whales predating on sea otters can determine nearshore benthic community structure because sea otters are considered a keystone predator in certain areas by controlling sea urchin populations and hence macrolagal communities (Estes *et al.* 1998).

Competition is an important “bottom-up” biotic factor controlling community structure. Space is often limiting in hard substrate environments and competition for this resource can be seen at the recruitment level. While timing, dispersal, growth rates, morphology, and other factors will be important in determining the winners in many competitions, many abiotic factors will assist in this determination, such as sedimentation and light. Competition for food and shelter can also be important in structuring nearshore rocky communities.

Abiotic factors can have an important role in determining structure in nearshore rocky communities. One of the most important factors is the presence and type of hard substrate. How stable and patchy hard substrate

is will determine community structure. For example, an intertidal bedrock community will be very different than an intertidal boulder community simply because disturbance caused by storm surge and waves will have a much larger impact on the more unstable boulder community. Water motion caused by storm surge and waves is a significant controlling factor structuring nearshore communities.

Temperature is another controlling factor for the subtidal, and particularly the intertidal zones. In the intertidal zone, desiccation caused by heat and freezing associated with cold snaps can be detrimental to both algal and invertebrate populations while they are exposed to air. Over large geographic areas in the subtidal, temperature will control species distribution. For example, the giant kelp *Macrocystis* is not found in subpolar areas while the bull kelp *Nereocystis* is not found in warm temperate areas. Similar species distributions along temperature gradients can be found in invertebrates and fish.

Other important factors include sedimentation, nutrients, contaminants and salinity. Of these, sedimentation can be particularly important because it has many cascading effects. Increased sedimentation can reduce light quality and quantity available for macroalgae, and it can also cause scouring and burial, which can be detrimental to the early settlement of many invertebrates and algae.

SUMMARY

Nearshore rocky shores create habitat and provide food, shelter and nursery grounds for numerous species, promote increased biodiversity, and enhance overall organism abundance, primarily because of the structural elements of macroalgae associated with them (Anderson 1994). Loss of these macroalgal areas could have profound impacts on regional patterns of biodiversity and ecosystem functioning, and ultimately on human social and economic interests through associated reductions in ecologically and commercially important species.

B. KONAR, UNIVERSITY OF ALASKA, FAIRBANKS, USA

Section 5 NaGISA Rocky Shore Protocol

First decide where to sample. Practical concerns of accessibility are a priority; if it is difficult to reach a site then future collection (in the years to come) may be compromised. Sites should be located in relatively undamaged locations that are free of any specific threats i.e. scheduled construction. Care should be taken to make sure that the site is not an anomaly in the general character of the seascape and that the rocky shore habitat extends beyond the immediate site. The trade offs between selecting previously sampled areas or new locations (historical time lines vs. exploring the undocumented) and how closely related your area is to other NaGISA sites are also factors that should be considered when contemplating a new site location. Finally it is a good idea to visit a site before committing to it as you will want to assess the practical issues of getting in and out of the water, determine high, mid and low tide levels and see how far out 1m, 5m, 10m and the optional 15m and 20m depth contours lie.

Once the decision of where to sample has been made, carefully read through the protocols, including the instructions on how to handle samples, confirm the availability of all the necessary equipment for the field and sorting stages and consult a tide schedule for the specific location. NaGISA Rocky Shore sampling is done both intertidally and subtidally and so is best done during a period of very low tides.

MATERIALS NEEDED: Transect tape (30m), 1 × 1m quadrat, 50 × 50cm quadrat, 25 × 25cm quadrat, labeled plastic bags and rubber bands, a scraper, a hand-held GPS unit, a thermometer, a field notebook, a pencil, a digital camera, a salinometer*, a light gauge*, (for underwater sampling add SCUBA gear, 63µm and 500µm mesh bags and underwater housing for the camera).

** Optional*

PRE SAMPLING: Label all collection bags and prepare datasheets including random numbers for all replicates.

SAMPLING:

1. Measure and record the surface water temperature and salinity at the site and the light intensity and temperature of each underwater depth (–1m, –5m, –10m, –15m, –20m).
2. Choose a starting point at one of the pre-selected heights/depths (see Fig. 5.1). Lay out the tape measure for 30m from this point along the same tidal height/underwater depth contour.
3. Find the location of the first replicate along this line (the first of your five previously chosen random numbers). Record the GPS of this point.
4. Place the 1 × 1m quadrat at the replicate mark along the tape measure.
5. Facing the water, place the 50 × 50cm quadrat at the lower left corner of the 1 × 1m quadrat and the 25 × 25cm quadrat in the bottom right corner of the 50 × 50cm quadrat (see Fig. 5.2).
6. Photograph the quadrats. Use the full frame and avoid shadows (label your photos to ensure that they can be identified later).
7. Assess the percent coverage of the different species of macroalgae in your 1 × 1m quadrat, count the *stipes* of the larger kelps and count all the macrofauna that is larger than 2cm. Record this information on a field datasheet.
8. Take voucher specimens of all the species; place them in correctly labeled plastic (or 500µm mesh underwater) bags. The label should

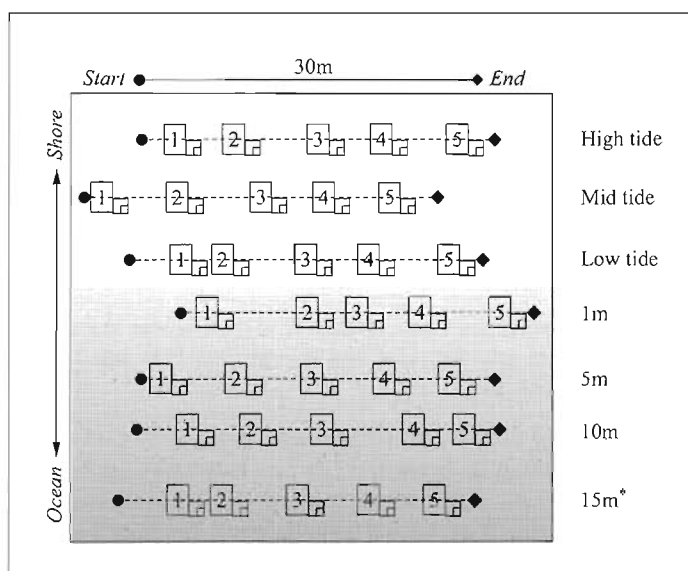


Fig. 5.1 Rocky Shore Layout

Here seven 30m transects are shown with five random replicates (each showing a complete quadrat set). Since replicates along each transect are chosen randomly, replicate numbers between transects may not line up. The 15m & 20m (which is not shown) depth contours are optional but official sites should have all other depths/heights.

- indicate that it is a voucher from the $1 \times 1\text{m}$ quadrat followed with the date, name of the site, tidal height and replicate number. Put the bag in a cooler as soon as possible.
9. Carefully remove all the macroalgae from the $50 \times 50\text{cm}$ quadrat. Place them in a correctly labeled plastic (or $500\mu\text{m}$ mesh underwater) bag. The label should indicate the quadrat size, date, site name, tidal height and replicate number. Put the bag in a cooler as soon as possible.
 10. Carefully remove all the material from the $25 \times 25\text{cm}$ quadrat. Place it in a labeled plastic bag (or $500\mu\text{m}$ mesh underwater). The label should indicate the quadrat size, date, site name, tidal height and replicate number. Put the bag in a cooler as soon as possible.
 11. Repeat steps 3–10 for each replicate along a height/depth line.
 12. Repeat this for high, mid and low tide heights and at -1m , -5m , -10m , -15m^* and -20m^* underwater. (* = optional)

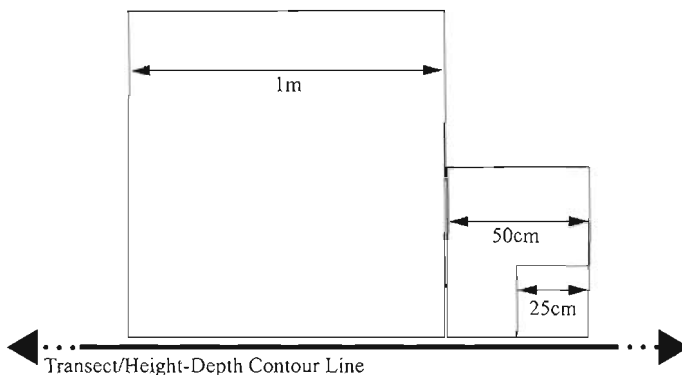


Fig. 5.2 Rocky Shore Quadrat Set

Instructions: Digitally photograph all 3 quadrats. Record percentage coverage/individual number of visible ($>2\text{cm}$) species in the $1 \times 1\text{m}$ quadrat. Collect all removable macroalgae from the $50 \times 50\text{cm}$ quadrat. Collect everything (by scraping) from the $25 \times 25\text{cm}$ quadrat.

FLOW CHART OF ACTIVITY

Coming in from the field you will have a digital camera full of photos, a handful of datasheets and coolers filled with sample bags. The photos must be downloaded onto your computer and labeled with their corresponding location, height/depth, replicate number and quadrat size. A CD library of the photos will enhance your reference collection, assist in identification and provide immediate viable comparisons for you and future researchers. The field data sheets (see section 23) must be combined and entered into the NaGISA excel data sheet (obtainable from your regional coordinator), which can be uploaded to the NaGISA website with preliminary data entered (and updated later). The sample bags must be carefully sieved and sorted into distinguishable groups as soon as possible.

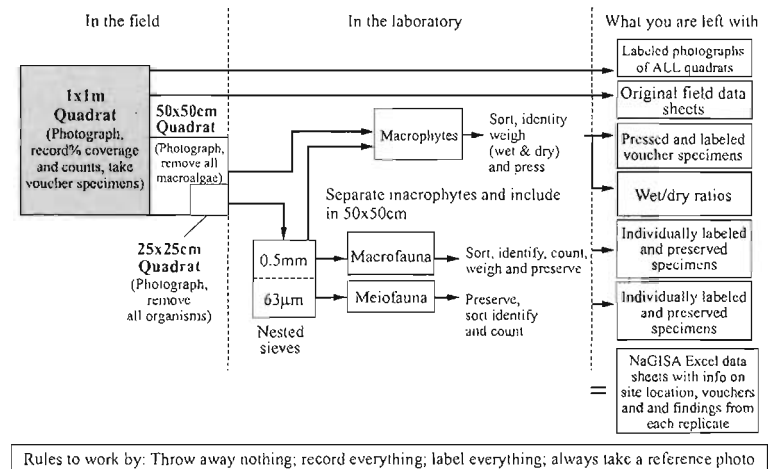


Fig. 5.3 Macroalgae: Activity Flow Chart

SORTING THE MATERIAL

- Each replicate is unique and must be dealt with separately.
- Wash the contents of the 25 × 25cm quadrat sample over 0.5mm and 63µm sieves (either nested or one after the other, with out losing material in the process).
- The material retained on the 63µm sieve will largely comprise of meiofauna and can be preserved directly and sorted later (see section 17).
- The macrobenthos in the 0.5mm sieve should be sorted into 'plant' and 'animal' and detritus, empty shells etc. removed.
- It is recommended that you sort, identify and weigh the macrophytes as soon after coming in from the field (the same day if possible). Remember to combine the macrophytes from the 50 × 50cm quadrat and the 25 × 25cm quadrat after the ones from the 25 × 25cm have been well cleaned over the nested sieves.
- For each macroalgal species with a sample weighing over 1g (otherwise it is simply marked as present) a wet weight – dry weight ratio should be established by drying a sub-sample of known weight of each species at 60°C for 24h. Selected samples should be pressed and voucher specimens made. Note the creation of pressing has priority over the establishment of wet and dry weight.
- The macrofauna should be sorted. Individuals should be counted and the wet weight determined for the species. Vouchers should also be made from these samples.

INTRODUCTION TO SORTING AND HANDLING SPECIMENS

Organizing and sorting are critical steps in the research process; the documentation of evidence and the establishment of a tracking method are particularly important to the quality of the resulting data. In the case of ecological fieldwork, as conducted by NaGISA with the main objective to determine species abundances the correct identification of specimens is a

priority. Regional keys are usually not enough to name specimens because there is no guarantee that they cover all available taxa. Furthermore, there is a lack of continuous update in the taxonomy of many groups. The best alternative is to use your voucher specimens to establish a reference collection either 'in house' or with the assistance of a museum or herbarium. This step should not be over looked, as reference to properly curated material is essential for all studies.

Museums or herbariums often publish procedures for preservation and long-term storage of the specimens in their institution; these are often related to the features of taxonomic groups (Green and Lambert 1994; Rose and de Torres 2002; Rose *et al.* 2002; Metsger and Byers 2003). In the following sections Part III and IV we outline specific details for handling targets groups that have been prioritized for identification within the NaGISA project.

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Section

6

Seagrass Bed Ecology

INTRODUCTION

In the coastal soft bottom areas, seagrass beds are an ecologically important habitat. The beds occur from the intertidal zone down to the subtidal. Their communities are highly productive, with a high degree of complexity and biological diversity. Seagrasses are submerged marine flowering plants that, unlike seaweeds, have stems, leaves, and rhizomes and produce flowers, fruits and seeds for their propagation. They provide three-dimensional systems with a network of underground structure (rhizomes) stabilizing the substrate condition in coastal areas. With these conditions, the beds can form several types of microhabitats for associated organisms. Hemminga and Duarte (2000) noted that animals associated with seagrass systems belong to three broad categories: infaunal species living within the sediments; epifaunal species living on the plants and the sediment surface; and epibenthic large mobile animals moving freely under and over the leaf canopy. Unlike coral reefs, salt marshes and mangroves, seagrass beds exist in nearly all coastal areas of the world, from tropical region up to the Arctic. Worldwide there are about 60 species of seagrasses (Short *et al.* 2001). They have a wide range of plant size with scale from a few centimeters (for example, genus *Halophila*) to a few meters (for example, genera *Posidonia*, *Phyllospadix* and *Enhalus*) (Fig. 6.1). Also, there are different morphologies among the different seagrass species. The different species and species number of seagrasses are, therefore, a major factor controlling the characteristics of seagrass communities and their productivity and diversity.

DIVERSITY AND DISTRIBUTION OF SEAGRASSES

Seagrasses are generally assigned to five families with 12 genera (Cook 1996). There are differences in generic distribution among geographical regions. In temperate including arctic regions, the five dominant genera are *Phyllospadix*, *Amphibolis*, *Posidonia*, *Zostera* and *Ruppia*, whereas the seven dominant genera in tropical regions are *Enhalus*, *Thalassia*, *Thalassodendron*,

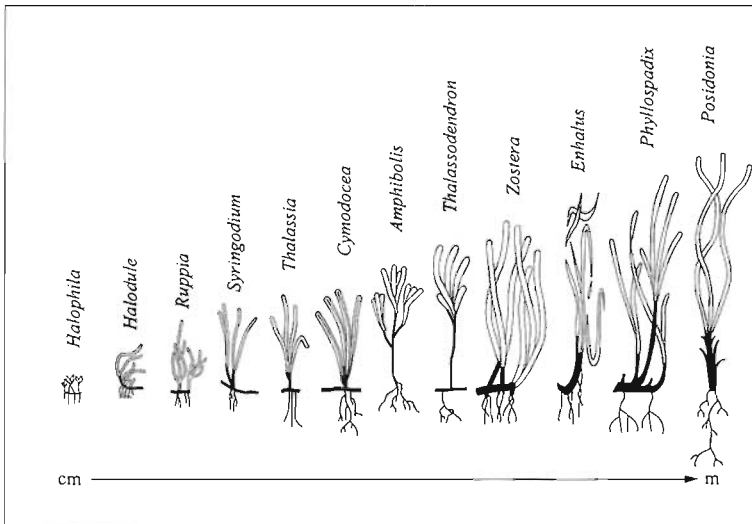


Fig. 6.1 Model of Seagrass Genera Arranged by Size

Syringodium, *Cymodocea*, *Halodule* and *Halophila*. However, a few genera overlap in geographical extent, e.g., *Zostera capricorni* extends into tropical Northeast Australia and *Ruppia maritima* is widely distributed throughout tropical regions, while some species of tropical genera such as *Cymodocea nodosa* also exist in temperate regions. Among ten regions identified for seagrass distributions, the highest species richness (24 species) was found in the Indo-West Pacific region while the lowest (one species, *Ruppia maritima*) was reported in South Africa (Short *et al.* 2001).

Typically, temperate and arctic seagrasses form monospecific beds. In northern temperate regions, *Zostera marina* is the most widespread and dominant species in both the Atlantic and Pacific Oceans, while *Posidonia oceanica* dominates Mediterranean seabeds. Also, other seagrass species, e.g. *Zostera japonica*, *Ruppia maritima* and *Phyllospadix* spp., can form characteristic beds. *Amphibolis* beds are unique in the temperate regions of the southern hemisphere. In tropical regions, seagrasses often form mixed-species beds. Commonly, beds in shallow waters less than 10 m deep are formed by various seagrass species, e.g., *Enhalus acoroides*, *Halophila ovalis*, *H. minor*, *Cymodocea serrulata*, *Halodule uninervis* and *Halodule pinifolia* in Thalen Bay, Krabi Province, Thailand. *Halophila decipiens* and *Thalassodendron ciliatum* are found in the seagrass beds from the intertidal down to the low subtidal zone and in the shallow waters surrounding the islands. In the eastern part of Indonesia and the southern and western coasts of the Philippines, *Thalassodendron ciliatum* is found down to 17 m depth (UNEP 2004).

ECOLOGICAL SERVICES OF SEAGRASS BEDS

Seagrasses form the basis of many complex coastal ecosystems and play a significant role as nursery ground by providing food, shelter and habitat for many groups of animals, such as fishes, crabs and shrimps. Juvenile stages of a number of fish species, some of them economically important, have been found to use seagrass beds before leaving to other habitats.

In northern Queensland, Australia, Cole *et al.* (1993) reported 134 fish species, most of them juveniles with an average length of 32 mm, in a tropical estuarine seagrass bed. Among the number of fish species spending their juvenile life stage in seagrass beds, economically important are, e.g., Atlantic cod (*Gadus morhua*) in eelgrass beds (*Zostera marina*) (Gotceitas *et al.* 1997), and at least 15 species of rabbitfish (*Siganus* spp.) in mixed species seagrass beds in the South China Sea region (UNEP 2004). Moreover, in Lombok Island, Indonesia, juveniles of many commercial species belonging to the genera *Siganus*, *Lethrinus*, *Lutjanus*, *Plotosus*, *Hemirhamphus*, *Tylosurus*, *Mugil*, *Caranx* and *Sphyrna* were recorded from monospecific seagrass beds of *Enhalus acoroides* and from mixed-species seagrass beds of *Cymodocea* and *Syringodium* (Hutomo and Peristiwady 1996). Regarding crustacean communities, many young stages of various commercial species exist in the seagrass beds, e.g. blue crab (*Callinectes sapidus*) in mixed seagrass beds (*Zostera marina* and *Ruppia maritima*) in Chesapeake Bay, USA (Orth *et al.* 1996), blue swimming crab (*Portunus pelagicus*) and various species of penaeid shrimp in mixed-species seagrass beds in Australia (Loneragan *et al.* 1994, Fortes 1995) and in Pakhlok Bay, Phuket, Thailand (unpublished data). For a detailed review of the role of seagrass beds as nursery ground see Hemminga and Duarte 2000.

Moreover, seagrasses play a vital role in marine food web dynamics. The leaves and stems of seagrasses support numerous epiphytic organisms. Sessile forms include benthic diatoms, small seaweeds, bryozoans, ascidians and sponges, and mobile forms include amphipods, tanaeids, isopods, harpacticoid copepods and nematodes. Small animals such as shrimps and snails graze upon these minute organisms. Subsequently, larger fish, shrimp and crabs eat these animals. Furthermore, in tropical regions, seagrasses are a main food for large animals, such as sea cow (*Dugong dugon*) (Erftemeijer *et al.* 1993, Marsh *et al.* 2002) and green turtle (*Chelonia mydas*) (Hemminga and Duarte 2000).

The presence of commercially important marine resources in seagrass beds is important for human subsistence in many coastal areas. Local fishermen can catch fish, shrimp and crabs by hand nets, beach seines and gill nets. Also, during low tide, they collect shellfish, sea cucumbers, sea urchins and seaweeds for their consumption and subsistence income. Fortes (1995) estimated the monetary values of seagrass beds as annual shrimp fishery values of about Aus\$700,000 in North Queensland and Aus\$1.2 million in Cairns Harbor seagrasses.

SUMMARY

Seagrasses are submerged marine flowering plants that, unlike seaweeds, have stems, leaves, and rhizomes and produce flowers, fruits and seeds for their propagation. They form highly productive and diverse communities on the soft bottom of all coastal areas of the world. There are about 60 seagrass species known worldwide. Temperate seagrass species often form monospecific seagrass beds, whereas in the tropical regions mixed species beds are more common. Obviously, seagrass beds play important ecological roles as nursery grounds and habitats for various commercial marine resources. Moreover, the beds are significant for the effective conservation of endangered species, such as the sea cow, in tropical regions.

Section

7

NaGISA Seagrass Protocol

Seagrass sites, like macroalgal sites, should be located in relatively undamaged accessible locations that are not scheduled to come under anthropogenic threats in the foreseeable future. Care should be taken to make sure that the site chosen for sampling is not an anomaly in the general character of the regions seagrass habitat i.e. the choice of where to sample should be made with a full understanding of the general region and the seagrass habitat found in the area. Specifically care should be taken to assess whether the region hosts; (a) intertidal and/or subtidal seagrass, (b) permanent fixed beds or small transitional beds and (c) mixed stands or homogeneous beds.

Once the decision of where to sample has been made, the protocols including the instructions on how to handle the samples should be read carefully and all the necessary equipment for the field and sorting stages prepared. At an appropriate time in the tidal cycle, sampling should be carried out in the middle of small single species beds or in horizontally distributed or mixed stands at 1m depth intervals throughout the bed.

While in small single species stands care should be taken to procure samples from the center of the stand (and optionally from the edges). Mixed stands or horizontally distributed beds require more thorough measurements (transects at 1m depth intervals) so that the variability of the habitat can be recorded. In general, the seagrass bed should extend beyond the transects, however as some areas naturally have very small individual beds this will not always be possible.

MATERIALS NEEDED: 50×50cm square quadrats, 15cm (inside diameter)×10cm (subsurface depth) corer, 2cm (inside diameter)×10cm (subsurface depth) corer, labeled plastic bags, a spade, a hand-held GPS unit, a thermometer, a field notebook, a pencil, digital camera, a salinometer, a light gauge*, (for underwater sampling add SCUBA gear, 63µm mesh bags and 500µm mesh bags and underwater housing for the camera).

** Optional*

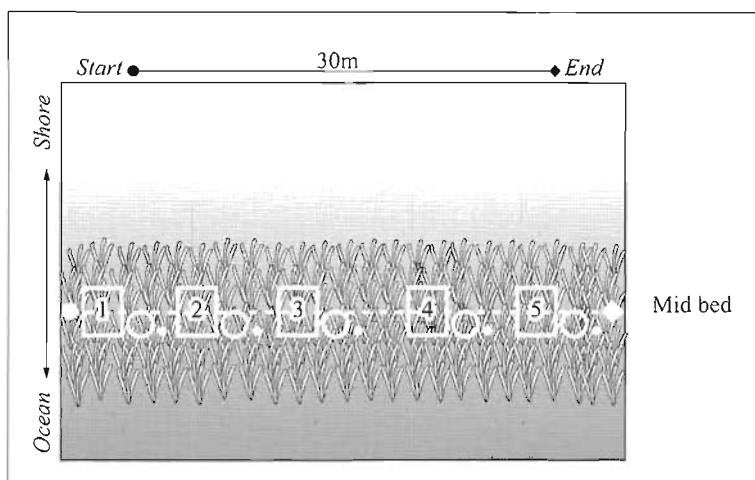


Fig. 7.1 Seagrass Bed Transect Layout

A 30m transect shown going through the middle of the seagrass bed, with five randomly placed replicate sets.

PRE SAMPLING: Before sampling, label all collection bags and prepare datasheets including random numbers for all replicates.

SAMPLING:

1. Measure and record the water temperature and salinity at the surface and at transect depth, measure and record the light intensity at the surface and at 1m intervals until reaching the lowest depth obtainable within the bed.
2. Choose a starting point for the main transect line so that it runs through the middle of the seagrass bed (Fig. 7.1). Record the height/underwater depth. Use a tape measure to mark off 30m from this point along the same depth contour.
3. Go to the first previously chosen random number along this line. Record the GPS of this point.
4. Place the 50×50cm quadrat at this point along the transect line.
5. Photograph the quadrat, taking care to use the full frame of the picture and getting no shadows in the picture (ensure that quadrat photos can later be labeled with their replicate number).
6. Count and identify all the macrofauna larger than 2cm taking voucher specimens as necessary. Be sure to put the vouchers in correctly labeled plastic (or 500µm mesh underwater) bags. The label should indicate that these are voucher specimens from the 50×50cm quadrat followed by the date, name of the site, tidal height and replicate number. Put the bags in a cooler.
7. Count the number of seagrass *shoots* within the 50×50cm quadrat. Record this information on the appropriate data sheet.
8. Place the 15cm core cylinder to the right hand side of the 50×50cm quadrat and place the 2cm core cylinder to the right of that larger core (see Fig. 7.2).
9. Push/twist the 2cm cylinder into the substrate to a depth of 10cm (Fig. 7.3). Place the cap on the 2cm corer and remove from the

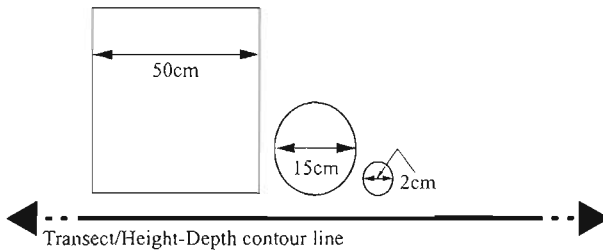


Fig. 7.2 Seagrass Bed Quadrat Set

Instructions: Photograph the 50×50cm quadrat, count and record the macrofauna (>2cm) and take voucher specimens, count the number of shoots in at least $\frac{1}{4}$ of the quadrat. Remove a 15cm diameter by 10cm depth core for a better assessment of the macrobenthos. Remove a 2cm diameter by 10cm depth core for meiofauna assessment.

substrate smoothly. Either cap the opposite end and place the whole core in a labeled plastic bag (or 63µm mesh underwater) or empty the contents into the labeled bag, rinsing the sides of the core with spray from a squeeze bottle. The label should indicate that it is the 2cm core sample followed by the date, name of the site, tidal height and replicate number. Make sure to get a full 10cm core – detritus and/or air bubbles in the sediment can prevent the full 10cm from being collected. If this happens repeat the core in a yet undisturbed place as close to the first one as possible. Keep the material damp and cool by placing the bag in a cooler or the mesh in a tub of seawater until it can be sieved and sorted.

10. Push/twist the 15cm cylinder into the substrate to a depth of 10cm (Fig. 7.3). Dig out from around the outside of the cylinder. Push the bottom cover under the cylinder and lift everything up together, empty the material into a labeled plastic (or 500µm mesh underwater) bag. The label should indicate that it is the 15cm core sample

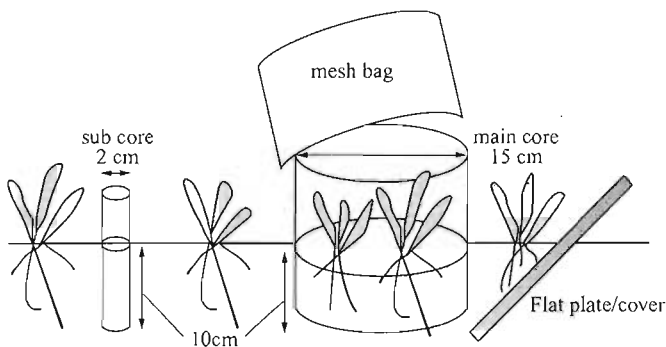


Fig. 7.3 Seagrass Bed Cores

Both the 2cm sub core and the 15cm main core are pushed/twisted into the substrate to a depth of 10cm. The sub core is capped before removing it from the substrate and wedge is used to help remove the main core. Covering the main core with a mesh bag prevents the loss of epifauna and reduces the loss of sample material.

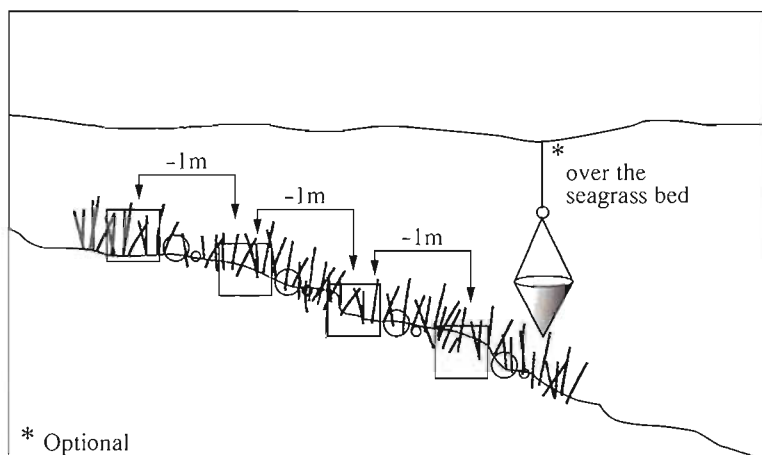


Fig. 7.4 Sampling Horizontally Distributed Seagrass Beds
Small single species stands should be sampled from the center of the stand (and optionally from the edges) and care should be taken not to sample between stands, but mixed stands or horizontally distributed beds are best sampled with multiple transects at 1m depth intervals.

followed with the date, name of the site, tidal height and replicate number. Remember to rinse down the sides of the cylinder with spray from a squeeze bottle. Keep the material damp and cool by placing the bag in a cooler or the mesh in a tub of seawater until it can be sorted.

11. Move on to the next random number along the transect line until all replicates are completed.
12. If the stand spreads over more than one depth contour then supporting transects placed at 1m depth intervals should be added as necessary, see Fig. 7.4.

FLOW CHART OF ACTIVITY

Coming in from the field you will have a digital camera full of photos, a handful of datasheets and coolers filled with sample bags. The photos must be downloaded onto your computer and labeled with their corresponding location, height/depth, replicate number and quadrat size. A CD library of the photos will enhance your reference collection, assist in identification and provide immediate viable comparisons for you and future researchers. The field datasheets (see section 23) must be combined and entered into the NaGISA excel data sheet (obtainable from your regional coordinator) which can be uploaded to the NaGISA website with even just the preliminary data entered (and updated later). The sample bags must be carefully sieved and sorted into distinguishable groups as soon as possible.

SORTING THE MATERIAL

- Each replicate is unique and must be dealt with separately.
- Wash the contents of the 15×10cm core over a 0.5mm sieve.
- Separate the seagrass and the macrofauna that is retained on the 0.5mm sieve.

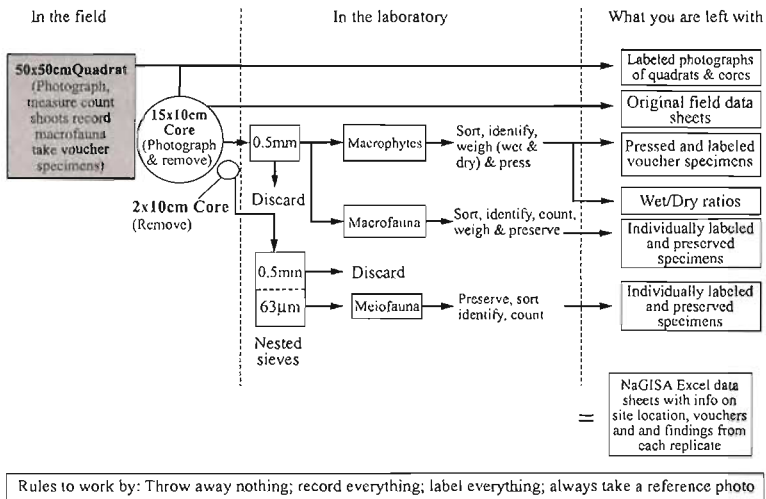


Fig. 7.5 Seagrass Bed: Activity Flow Chart

- It is recommended that you sort, identify and weigh the seagrass soon after coming in from the field (the same day if possible).
- Weigh the seagrass (species separately and establish a wet weight – dry weight ratio by drying a sub-sample of known weight of each species at 60°C for 24h. Selected samples should be pressed and voucher specimens made. Note the creation of pressing has priority over the establishment of wet and dry weight.
- The macrofauna should be sorted. Individuals should be counted and the wet weight determined for the species. Vouchers should also be made from these samples.
- Wash the contents of the 2×10cm core over 0.5mm and 63µm sieves (either nested or one after the other, without losing material in the process). Discard the material in the 0.5mm sieve. The material retained on the 63µm sieve will largely comprise of meiofauna and can be preserved directly and sorted later (see section 18).

INTRODUCTION TO SORTING AND HANDLING SPECIMENS

Organizing and sorting are critical steps in the research process; the documentation of evidence and the establishment of a tracking method are particularly important to the quality of the resulting data. In the case of ecological fieldwork, as conducted by NaGISA with the main objective to determine species abundances, the correct identification of specimens is a priority. Regional keys are usually not enough to name specimens because there is no guarantee that they cover all available taxa. Furthermore, there is a lack of continuous update in the taxonomy of many groups. The best alternative is to use your voucher specimens to establish a reference collection either 'in house' or with the assistance of a museum or herbarium. This step should not be over looked, as reference to properly curated material is essential for all studies.

Museums or herbariums often publish procedures for preservation and long-term storage of the specimens in their institution; these are often related

to the features of taxonomic groups (Green and Lambert 1994; Rose and de Torres 2002; Rose *et al.* 2002; Metsger and Byers 2003). In the following sections Part III and IV we outline specific details for handling target groups that have been prioritized for identification within the NaGISA project.

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Part III

Specific Details for Working with the Primary Target Groups

Section 8 Macroalgae



(photo courtesy of B. Konar)

CHARACTERISTICS OF MACROALGAE

- 1) Macroalgae are eukaryotic multicellular algae
- 2) Usually attached to substrate
- 3) Brown, red and green algae differ in their pigment composition, storage components, and cell structure
- 4) Complex life histories including single-celled dispersal stages (mostly flagellated)

INTRODUCTION

Taxonomic classification is continuously changing and information about the phylogeny of algae depends on the sources used. Traditionally, algae were placed in the kingdom Protista and consisted of Rhodophyceae (red algae), Ulvophyceae (green algae; formally called Chlorophyceae) and Phaeophyceae (brown algae). More recently (see www.algaebase.org), they have been placed in the kingdoms Plantae (Chlorophyta: green algae; Rhodophyta: red algae) and Chromista (Ochrophyta: brown algae). They are generally multicellular organisms that attach to substrates like rocks, shells

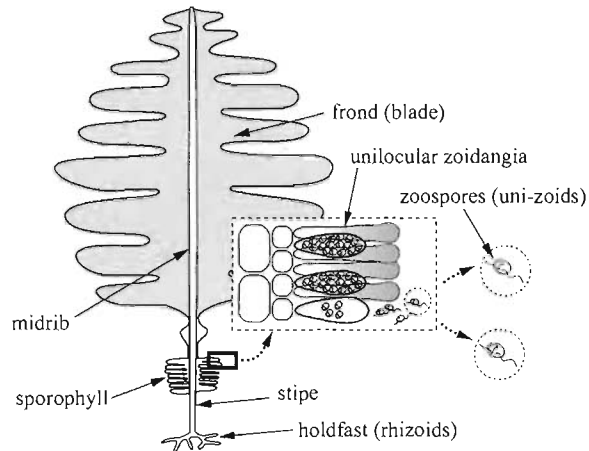


Fig. 8.1 General Morphology of Macroalgae

and other macroalgae with holdfasts (different from the roots of vascular plants). Some of them have gas bladders or pockets of air for buoyancy allowing them to stay relatively erect in the water column. The thalli (fronds, stipes and holdfasts) of brown macroalgae, such as Laminariales (kelps) often exceed several meters in length. Cool-temperate regions often harbor kelp communities that build multi-layered habitats for many associated smaller macroalgae and animals referred to as kelp forests. In contrast, tropical communities are usually comprised of small macroalgae that usually occur in multi-species clusters on reef flats. These so-called turf algae produce much less biomass but can be as high in species diversity as temperate kelp forests.

Macroalgae principally propagate by means of zooids (such as zoospores and gametes), which can be either flagellated as they are in brown and green algae or non-flagellated such as the tetraspores and carpospores of red algae. Macroalgae have varied life histories, which often include the alternation of generations. Sometimes, alternate life history stages have very different morphologies referred to as heteromorphic and which are presumably an adaptation to seasonal changes in the environment. Macroalgae are found from the intertidal zone to depths of over 100m with the highest species diversity and abundance occurring in the upper subtidal zone. The maximum depth at which algae can grow is limited by their need for light. The depth of the light compensation point differs depending on the species, but generally is in the range of 0.5 to several percent of water surface light intensity.

GENERAL MORPHOLOGY

The organization of macroalgal thalli is relatively simple. The thalli of brown and most red macroalgae are composed of a colorless medullary layer and a richly pigmented cortical layer.

Green algae are often simply organized and undifferentiated in their organization. In addition to the typical multicellular macroscopic forms, green algae can also exist as unicellular macroscopic forms. These consist of a single large cell with numerous nuclei and are called coenocytes or are referred to having coenocytic organization.

Macroalgae have no root systems or flowers and most of the reproductive structures are embedded in the algal tissue. They are normally formed near the surface layer and are visible on the surface as densely colored patches. Some brown algae develop specialized reproductive structures (receptacles, sporophylls). Different life history stages (i.e. gametophytes and sporophytes) may have remarkably different morphology. Even if the gross morphology is indistinguishable by external appearance, they form different types of reproductive organs on separate thalli.

Some taxa form crustose thalli tightly attached to substrates (e.g. non-geniculated coralline algae, ralfsoid brown algae). Some taxa (e.g. corallines and *Galaxaura* in red algae, *Padina* in brown algae, and *Halimeda*, *Acetabularia* and many others in green algae) are calcified, depositing CaCO_3 around or within cells.

PREPARING SPECIMENS FOR IDENTIFICATION

When collecting macroalgal specimens, the entire thallus (fronds, stems and holdfasts) must be collected, as identifying macroalgae based on fragmented specimens can be difficult. In many taxa, the anatomy of the reproductive structures is an essential character for identification, and so it is possible that for taxonomic purposes specimens may have to be collected outside of the NaGISA quadrats or even at a different time of the year so as to collect plants of different life history stages.

Collected specimens should be observed when fresh and special note taken of the following features: colour; length, width and thickness of the thallus; branching pattern; shape of erect thallus (is it terete, flat or foliose); shape of holdfast (is it cupped or knotted); presence/absence of gas bladders (are they hollow or solid); tissue anatomy (are the surface and inner layers differentiated); reproductive structures when present; presence/absence of special non-reproductive structures such as hairs, glands, ascocysts); presence/absence of meristematic cells/regions.

Not all the macroalgae from the 50 × 50cm quadrats need to be preserved but ample examples and vouchers of each species should be preserved. Pressing macroalgae is the most common method for preservation and pressed specimens are suitable for later morphological and anatomical analysis.

Pressing: Place fresh wet specimens on herbarium mounting paper and using as little water as possible spread the thallus out, displaying the characteristic branching pattern. Cover with gauze or wax paper and press between absorbent sheets of paper. Bind specimens together in a herbarium press dry in a well-ventilated area. NaGISA protocols require that voucher specimens be pressed (see glossary of methods for greater detail) but the following methods can complement this technique:

Silica gel: Place the algae in an airtight container covered with silica crystals. Specimens treated like this are suitable for DNA extractions although less so for morphological observations as they will shrink and change shape.

Formalin: Place specimens in a jar with 4% neutralized formalin in seawater (see glossary of methods). Specimens treated like this are suitable for later anatomical observations, but the thallus will lose its colour; DNA extractions from formaldehyde-preserved specimens have not been very successful. Formalin is toxic, and this method requires a relatively large storage space.

Freezing: Place the specimen in plastic bag, if possible laying it out flat. Frozen specimens are suitable for DNA extraction and morphological observations and will not lose color but consume considerable space and energy, and risk damage by mechanical failure of freezers or by power interruptions.

Macroalgal guidebooks are usually specific to a region and taxonomic keys are helpful, especially in the case where the local guidebooks may not cover a specific area or may be out of date. The use of taxonomic keys requires basic knowledge of the local macroalgal flora of and morphology to avoid misidentification, beginners should assign specimens to higher taxa before attempting to identify specimens to species level. Always start with a field guide with color illustrations to make a preliminary guess based on the gross morphology, and then make a more detailed anatomical examination. Some red and brown algae have similar external appearances, but can easily be distinguished by the tissue structure, chloroplast color, and reproductive structures.

For the taxonomy of brown algae, mode of growth (presence/absence of meristematic cell(s)/zone), number of chloroplasts and the presence/absence of pyrenoids, and morphology of reproductive structures are important characters at the ordinal level. For the taxonomy of green algae, basic tissue structure (filamentous or membranous when multicellular; giant cells or composed of tubular structures when coenocytic), morphologies of reproductive structures and zooids are important ordinal characters. For the taxonomy of red algae, presence/absence of pit plugs (structures connecting adjacent cells), ultra-structure of pit plugs, morphology of reproductive structures (especially the developmental patterns of carposporophytes), and basic architecture of erect thalli are important ordinal characters.

To observe tissue anatomy hand-section specimens using a light microscope: 1) prepare a small piece of the thallus (5–10mm × 10–20mm); 2) put the tissue on a glass slide; 3) place another glass slide on the tissue; 4) slice the tissue with a razor blade using the edge of the top slide as a guide, and slide the top slide gradually after each cut; use forceps to place the thinly-sliced tissue on a glass slide, add several drops of seawater cover with a cover-slip and observe under a microscope. Before sectioning coralline algae decalcify it by adding a few drops of 1N HCL (allow to it to bubble).

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Section 9 Seagrasses



(photo courtesy of L. Benedetti-Cecchi)

CHARACTERISTICS OF SEAGRASSES

- 1) Flowering plants with roots, stems and leaves
- 2) Shoots arising from rhizomes
- 3) Leaves strap-like or ovate, with air channels
- 4) Mostly in shallow (<50m) soft-bottom environments

INTRODUCTION

Seagrasses are submerged marine flowering plants. Unlike seaweeds they have stems, leaves, rhizomes, roots and produce flowers, fruits and seeds. They grow well in various types of sediment, namely sand, mud, fragmented coral and shell and some species even grow in rocky habitats. They are found in the intertidal to the subtidal zone from the equator to boreal regions. In coastal areas they build complex and productive habitats, called seagrass beds. Many species of fauna and flora exist in this habitat either permanently or temporarily, creating highly diverse communities.

GENERAL MORPHOLOGY

Seagrasses are structurally similar to terrestrial grasses. The plants consist of two main parts: the aboveground portion; stems, leaves and reproductive structures and belowground structure; rhizomes and roots (Fig. 9.1). While most seagrasses are similar in general shape, several different morphological

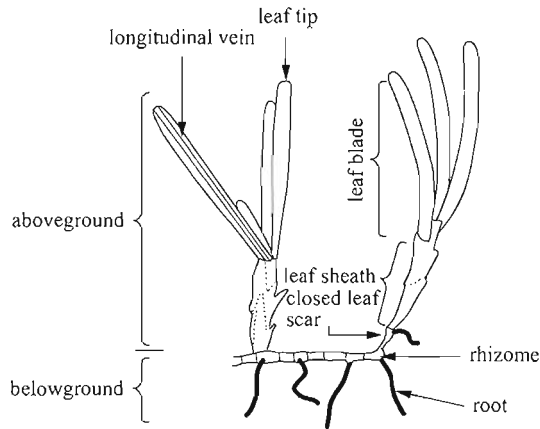


Fig. 9.1 Key Morphological Characteristics of Strap-Like Leaf Blade Seagrasses

forms of seagrasses can be distinguished. The shape of the leaf blade is a key character for seagrass identification. Mainly, there are two types of leaf shape: strap-like (Fig. 9.1) and oval to oblong (Fig. 9.2). Major morphological parts of seagrass plants are explained here briefly:

Leaf: the most obvious part with useful features for identification, such as tip, veins and sheath. *Leaf tip* is the end part of a leaf blade and can be rounded or pointed. *Leaf veins* are strands of vascular tissue in the leaf, to transport water, nutrients and photosynthetic products, and their pattern, direction, placement and number are used for identification. *Leaf sheath* is the lower portion of a leaf claspings the stem in seagrass with strap-like leaf blades. When the abundance of seagrasses is investigated, this part is counted and expressed as number of shoots per m². *Petiole* is a stalk of a leaf in seagrass with oval shaped leaf blade. *Leaf scar* is a scar on a stem or rhizome marking where and how (closed or opened) a leaf was once attached.

Flower: a reproductive organ surrounding reproductive seeds. Seagrasses are dioecious, with separate male and female flowers on different plants, or monoecious, containing male and female flowers on the same individual plant. Also, hermaphrodite flowers, combining functional male and female parts in the same flower, are found in some seagrasses.

Rhizome: a horizontal axis of a seagrass plant, commonly within sediment, it is often a major part of the underground plant biomass. It is formed in segments. Leaves or vertical stems arise from the joints of the segments, called *nodes*. The interval between nodes is called an *internode*.

Root: underground tissues growing from a node with the function of nutrient uptake and stabilization of the plant. Size and presence of root hairs are used for identification.

PREPARING SPECIMENS FOR IDENTIFICATION

Seagrasses should be collected as whole plants because both aboveground and belowground portions may be necessary for identification. The corer described in the protocols section should be able to do this effectively but

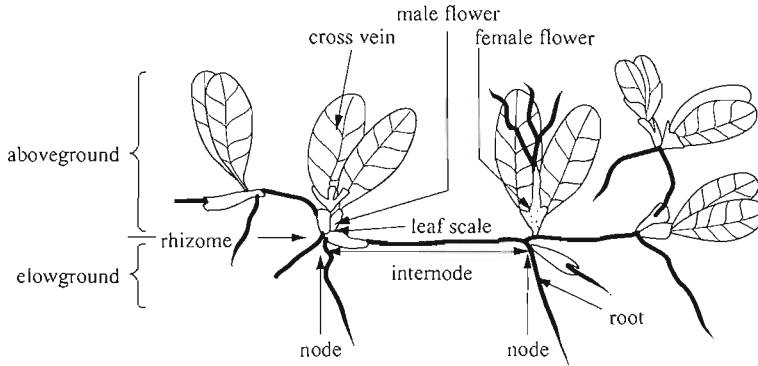


Fig. 9.2 Key Characteristics of Oval Shaped Leaf Blade Seagrasses

if you are worried that you do not have a complete specimen gently wedge a trowel or shovel deep underneath a desired specimen. Back in the lab debris can be washed off the specimens over a coarse sieve 500µm and a pair of forceps is useful to aid in removing bits of shell, gravel and epiphytic organisms. Fresh specimens are best kept moist in plastic bags or bottles. If specimens cannot be examined immediately, they may be stored for a few days in the vegetable compartment of a refrigerator. For longer maintenance, specimens have to be preserved in 4% neutralized formalin in seawater (see glossary of methods), and although seagrass can be preserved wet long-term preservation is best gained by drying specimens in a plant press (see glossary of methods). Well-dried specimens should be labeled with sampling information, such as species name, collector name, sampling place and date of collection and wrapped with a plastic sheet or put in a paper holder to protect them from dirt.

DIVERSITY

Seagrasses belong to the subclass Monocotyledoneae within the class Angiospermae. These marine plants are divided into five families: Posidoniaceae, Zosteraceae, Ruppiaceae, Cymodoceaceae and Hydrocharitaceae. The first two families are predominantly temperate, while the last two are primarily found in the tropics. The family Ruppiaceae is found both in temperate and in tropical regions and mostly in low saline water. There are 13 genera found within the five seagrass families: one Posidoniaceae (*Posidonia*); three Zosteraceae (*Zostera*, *Heterozostera* & *Phyllospadix*); one Ruppiaceae (*Ruppia*); five Cymodoceaceae (*Cymodocea*, *Halodule*, *Syringodium*, *Thalassodendron* & *Amphibolis*) and three Hydrocharitaceae (*Enhalus*, *Thalassia* & *Halophila*). A description of each genus follows:

Genus *Posidonia*: Five species (four found in Australia and one in the Mediterranean region). Strap-like leaf blades, leaves bearing a ligule at junction of sheath and blade, leaf sheath persists as bundle of fibers, monoecious; spike inflorescence with various numbers of hermaphroditic flowers.

Genus *Enhalus*: one species (*Enhalus acoroides* from the east coast of

Africa to the Indo-West Pacific). The largest tropical seagrass, strap-like leaf blade, non-ligule leaves with a basal sheath persisting as fibers, dioecious; small white male flowers in inflorescence with submerged short peduncle; female flower with white petals enclosed in brown spathe sheaths and attached on long peduncle.

Genus *Thalassia*: two species (*T. hemprichii* from the east coast of Africa to the Indo-West Pacific and *T. testudinum* in the tropical western Atlantic). Medium-sized seagrasses, strap-like leaf blade, non-ligule leaves with a basal sheath not persisting as fibers, thick rhizome prominently marked by several shoot closed scars, dioecious; pedunculate inflorescence with one to two flowers on the male plants and one on the female plants.

Genus *Halodule*: six species (wide distribution along the coasts of all tropical seas). Slender strap-like leaf blade, leaves bearing a ligule and with numerous tannin cells, having one to three longitudinal veins, leaf tips with pointed, bi- or tri-dentate feature, unbranched roots, dioecious, very obscure flower forming in the base of the leaf sheath, fruit with few seeds at the base of the shoot and having a seed bank.

Genus *Cymodocea*: four species (*C. serrulata* & *C. rotundata* in the Indo-Pacific, *C. nodosa* in the Mediterranean and *C. angustata* in western Australia). Strap-like leaf blade, leaves arising from vertical stems, with ligule and numerous tannin cells, seven to 17 longitudinal veins, rounded or serrated leaf tip, branched roots, dioecious, sessile or shortly stalked female flower with an ovoid ovary and stalked male flower with two anthers, mature seeds with dark coloured, hard coated, beaked nut with various ridges along the length.

Genus *Thalassodendron*: two species (*T. ciliatum*: wide distribution in the tropical part of the Indo-Pacific and *T. pachyrhizum*: in western Australia). Strap-like leaf blade with spinulose margin, leaves bearing a ligule with numerous tannin cells, stem not at each rhizome node, one unbranched or sparsely-branched erect stem on every fourth rhizome node, dioecious, solitary male or female flower attached on short lateral shoots, false fruits.

Genus *Amphibolis*: two species (*A. antarctica* and *A. griffithii*: wide distribution along the southern and western coasts of Australia and on Tasmania). Similar morphological characteristics to those in genus *Thalassodendron* but with entire margin of leaf blade, one profusely branched erect stem every four to eight rhizome internodes.

Genus *Zostera*: ten species (wide distribution in the northern Pacific, the northern Atlantic with the best known species, *Z. marina* (eelgrass) and few species (*Z. muelleri* & *Z. capensis*) in the tropical and subtropical Australia). Strap-like leaf blade, leaves bearing a ligule but without tannin cells, rhizomes not congested, with long (more than 2 mm) internodes, monopodial and herbaceous rhizome with a short lateral shoot at each node, hermaphroditic monoecious, distinguish spadix inflorescence containing male and female flowers enclosed within leaf spathe at the base of the leaf blade and attaching on a specialized shoot, fruit ovoid or ellipsoid, small nut-like seed with hard coat.

Genus *Heterozostera*: one species (*H. tasmanica* found from the western Australia to New South Wales, Australia, also in Tasmania and Chile). Similar morphological characteristics to those in genus *Zostera* with

differences in having a sympodial and ligneous rhizome with an erect unbranched, deciduous shoot at each node.

Genus *Phyllospadix*: five species (wide distribution in the northern part of Pacific). Similar morphological characteristics to those in genus *Zostera* with the differences being short compacted internodes (1–2 mm long), dioecious, spadix linear lanceolate containing various numbers of male (male spadix) or female flowers (female spadix), crescent shaped fruit with a soft exocarp and a hard fibrous endocarp.

Genus *Syringodium*: two species (*S. isoetifolium* in the Indo-Pacific and *S. filiforme* in the Caribbean). Terete-shaped leaf blade, leaves bearing a ligule, dioecious, flowers forming a complex inflorescence cymose (called a cyme), nut-like seed with beak and hard coat.

Genus *Ruppia*: few species (*R. maritima*: well known species in low salinity water). Fine and thread-like leaf blade circular in cross section, fragile rhizome with numerous roots, monoecious, male and female flowers in a single inflorescence with male flowers maturing early, small beaked drupe seeds and deposition of seed bank in sediment.

Genus *Halophila*: 11 species (the most diverse genus and wide distribution in all tropical areas, extending into subtropical and warm-temperate waters). Fragile round (oval to oblong) leaves with petioles and in pairs or attached on obvious vertical stem with more than two leaves, monoecious or dioecious, forming seed bank in some species.

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Section 10 Echinodermata



(photo courtesy of T. Kato)

CHARACTERISTICS OF ECHINODERMATA

- 1) Pentameric symmetry
- 2) Water vascular system
- 3) Stereom endoskeleton
- 4) Bilaterally symmetric larvae possessing rudiment structure and undergoing metamorphosis
- 5) Mutable collagenous tissue

INTRODUCTION

The Echinodermata (*echino* = “spiny”; *derm* = “skin”) is an entirely marine phylum of deuterostome macroinvertebrates with great morphological diversity. The echinoderms include forms such as the flower-like sea lilies and feather stars (Crinoidea), sea stars (Asteroidea) and brittle stars (Ophiuroidea), hard-bodied, spiny sea urchins (Echinoidea), and the soft-bodied and somewhat wormlike sea cucumbers (Holothuroidea). Most extant taxa show radial symmetry based on five rays (pentamery), though some forms have supernumerary rays. All echinoderms have a water vascular system made up of a central water ring that gives rise to five radial vessels that connect to the tube feet (podia). Tube feet are important external manifestations of this unique echinoderm character, figuring strongly in locomotion, feeding, respiration, and as sense organs. Rows of tube feet are arranged in rays that define regions and systems of plates called ambulacra. The water vascular system is connected to the surface of the animal via a stone canal leading to an external, sieve-like madreporite perforated by hydropores, although there sometimes is a secondary loss of this connection.

Most echinoderms have a well-developed internal skeleton of elements composed of polycrystalline calcium carbonate with a protein matrix called stereom. All the spines, plates, and other hard parts are made of stereom. It is important to note that this is not true shell. Even when arranged as a

hard, hollow casing as in echinoids (in which it is known as a “test”), it is an endoskeleton normally covered with epithelium. Echinoderms are also characterized by a two-part life history in which the bilaterally symmetric larvae, usually planktonic, develop a structure called the rudiment. Prior to metamorphosis, the rudiment initiates the development of the pentaradial symmetry of the adult. Echinoderms are also unique among invertebrates in possessing mutable collagenous tissues that can be stiffened or softened through nervous control and allow the animal to maintain a specified shape without expending muscular energy.

Other conspicuous features unique to echinoderms include spines and pedicellariae. The former can be articulated on a ball and socket joint and pointed or moved using a basal ring of muscles. Spines can also grow directly from the surfaces of plates and ossicles, without articulation. Pedicellariae are constructed of small, jaw-like, opposable pincers (valves) made of stereom. They are distributed over the surfaces of some echinoderms, but certain types can be highly regionalized. Pedicellariae are used to ward off predators and fouling organisms that might otherwise settle onto an echinoderm’s body. In asteroids, they are usually made of two jaws set low on the body, either directly upon the plate surface, or on a small ossicle. In echinoids, they assume a myriad of forms, and can be constructed of two or three jaw ossicles (valves) mounted on the ends of spine-like stems of varying lengths. In some instances there are as many as four or five of these valves. Pedicellariae figure prominently in many taxonomic works, notably Mortensen (1928–1951).

There are many rocky shore and seagrass species, but echinoderms also occur on sandy beaches and coral reefs as well as at the greatest depths of the sea. They range in lifestyle from pelagic larvae to infaunal burrowers and inhabit virtually every conceivable marine environment. However, as they do not have complete barrier between the internal water vascular system and ambient water, they cannot live in areas of very low salinity. Echinoderms have left a relatively complete and detailed fossil record over 500 million years long.

GENERAL MORPHOLOGY

The best way to approach the morphology of this phylum is to survey the representative groups and appreciate the important ways in which they differ. This helps to determine which resources to use for further identification.

Tube feet. External, sometimes suckered extensions of the water vascular system, usually found in rows along the body or arm to form an ambulacrum

Spine. Spikes of stereom that articulate with the body wall

Arm. An extension of the body wall accompanied by an ambulacrum

Disk. The central body at which the arms meet in brittle stars and sea stars (starfish)

Feeding tentacles; anterior extensions of the water vascular system used for feeding by sea cucumbers

Madreporite. Special, perforated plate on the top of sea stars, underside of brittle stars, and in the apical region of sea urchins that covers the external opening of the water vascular system

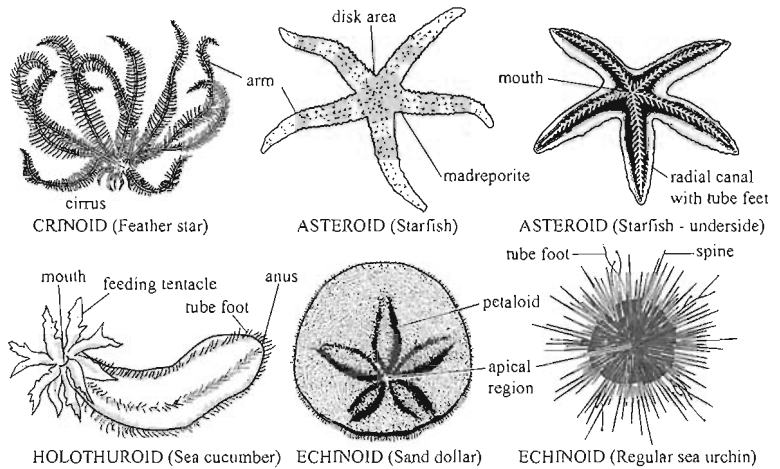


Fig. 10.1 Representative Echinoderms

Mouth. Usually centralized, on the underside of sea stars, brittle stars, and sea urchins, but on the top of the central cup-like body of feather stars, and at one end of the elongated body of sea cucumbers

Anus. Absent in brittle stars and many sea stars, at the top of the body in sea urchins, and at the opposite end of the body from the mouth in sea cucumbers

Petaloid. The flowerlike arrangements of specialized tube feet in irregular urchins such as sand dollars and heart urchins

The techniques used for identification are as diverse as the echinoderm groups. Characteristics used to key out the various taxa are highlighted below for each of the five main groups.

Crinoidea. Sea lilies are stalked forms and are generally rare in shallow water. Feather stars are common in tropical coral reefs, and vary in arm number and arm branching patterns. The number and form of cirri at the base of the cup, and the spination and form of pinnules along the arms are also used in many keys.

Asteroidea. The presence or absence of suckers on the tube feet, types and distributions of pedicellariae, and the morphology and number of plate series along the arms are important features. Many keys also use the ratio of major radius (center of the disk to the arm tip, "R") to minor radius (center of the disk to the inter-arm margin, "r") as a diagnostic character.

Ophiuroidea. Keys rely on plate patterns on the central disk, arm plating and spination, and the arrangement of small papillae and teeth on the jaw ossicles on the underside of the disk. Color, when it is preserved or recorded in photographs, can also be helpful.

Holothuroidea. Presence or absence of tube feet along the body, number and morphology of feeding tentacles, and most importantly, the shapes and

distribution of tiny ossicles in the body wall are used in sea cucumber taxonomy.

Echinoidea. Spination, color, plate architecture and overall shape of the test, number and types of pedicellariae, presence or absence of the jaw apparatus (Aristotle's lantern), arrangement of ambulacra, and placement of the anus within or outside the apical region are only some of the main features of sea urchins used in identification.

PREPARING SPECIMENS FOR IDENTIFICATION

Few echinoderms are dangerous, but some echinoids are best handled carefully, or with gloves. Some forms have poison glands at the spine tips but the danger from these can be greatly exaggerated. To preserve tube feet and the body form of soft-bodied taxa like sea cucumbers it is best to relax the specimens before fixing them, the use of magnesium chloride, magnesium sulfate, ethanol or cool water treatment (see glossary of methods) are all acceptable. Once the echinoderm has become sufficiently unresponsive it can then be injected with and placed in the fixative of your choice (see list below) with little or no change in the orientation of soft parts. Most echinoderms, excepting the sea cucumbers, lend themselves well to drying for long-term storage and manipulation for identification. The stereom lends rigidity, and helps the specimen to keep its form even after desiccation. However, the specimen should be fixed in 70% or stronger solutions of ethanol for several days before drying in a well-ventilated area out of direct sunlight. Dried sea urchins, sea stars, and brittle stars can be readily identified with most keys. Wet-preserved material is necessary for the soft-bodied sea cucumbers. As desiccated specimens can disarticulate over time, sea lilies, feather stars, brittle stars, and many of the more delicate sea stars are best preserved as wet material in >70% ethanol or 4% neutralized formalin in seawater.

To preserve soft tissue morphology and the overall body form of sea cucumbers the specimens must be wet fixed. This can be done with 4% neutralized formalin-seawater followed by transfer to 75% ethanol for long-term storage or for material that will be used for molecular studies, in 95% ethanol. For larger specimens, injection by syringe of the selected fixative/preservative is recommended. This is especially true for material to be preserved in ethanol rather than formalin because ethanol must be brought into contact with the internal organs as soon as possible.

Certain of the major groups require specialized techniques to view morphological structures for identification. For pedicellariae of asteroids and especially echinoids, the pedicellaria can be removed with fine forceps and placed in a drop of bleach (5% sodium hypochlorite in a small dish or microscope slide). Once the soft tissues are dissolved, the type and morphology of the jaw and stem ossicles can be observed. Similarly, small pieces of the body wall of holothuroids can be dissolved to reveal the ossicles. Monitoring of the tissue digestion process is recommended because the spatial orientation of the ossicles can also be informative. In echinoids, notably the irregular forms (sand dollars, heart urchins), the plate architecture of dried specimens can be revealed by brushing a solution of equal parts of 95% ethanol and glycerin onto the test once the test is lightly cleaned with a stiff toothbrush. As the ethanol evaporates, the glycerin darkens the plate sutures. For sand dollars, it is useful to polish the test with a graded series of sand papers before application of the solution.

DIVERSITY OF LIVING ECHINODERMS

The Phylum Echinodermata is divided into five classes as follows:

Crinoidea (sea lilies, feather stars) ~625 species, they possess a central body cup, or calyx, which is surrounded by feathery, usually heavily branched arms. Sea lilies have a stem, feather stars lack one.

Asteroidea (sea stars or starfish) ~1,500 species in five orders: Platysterida, Paxillosoida, Valvatida, Spinulosida, Forcipulatida and the debated sea daisies sometimes give their own class, Concentricycloidea. They typically have five unbranched arms radiating from a central disk, which is confluent with the arms themselves.

Ophiuroidea (brittle stars and basket stars) ~2,000 species in three orders: Oegophiurida, Phrynophiurida, Ophiurida. They typically have five arms, but the central disk is more clearly demarcated from the arms. Basket stars are a type of ophiuroid with branched arms.

Holothuroidea (sea cucumbers) ~1,150 species in six orders: Dactylochirotida, Dendrochirotida, Aspidochirotida, Elasipodida, Molpadida, Apodida. They have a cluster of tentacles around the mouth at one end, the anus at the other, and may or may not have tube feet in ambulacra along the body.

Echinoidea (sea urchins, sand dollars, heart urchins, lamp urchins), ~950 species in ~13 orders divided into a rather complicated series of subclasses, infraclasses and cohorts. They usually have a rigid body wall of sutured plates upon which are mounted a dense forest of spines.

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Section 11 Polychaeta



(photo courtesy of T. Kato)

CHARACTERISTICS OF POLYCHAETA

- 1) Bilateral symmetry
- 2) Segmented worms
- 3) Usually have a pair of non-segmented parapodia on each segment
- 4) Usually chitinous chaetae arising from parapodia
- 5) Abundant in most marine environments, with a few species found in fresh water and terrestrial environments

INTRODUCTION

Polychaeta is a group of segmented worms belonging to the phylum Annelida. Commonly known as seaworms or bristle worms they include a vast array of different groups; fire worms (Amphinomidae), scale worms (for example, Polynoidae, Sigalionidae), tubeworms (for example, Sabellidae, Serpulidae) etc. Like other members of the Annelida (such as Clitellata, including oligochaetes and hirudineans), the body of polychaetes consists of a longitudinal series of more or less similar body segments. A large number of polychaetes are easily distinguished from clitellates by having various well-developed appendages on their heads and other body parts; although some polychaetes have simple body plans lacking any appendages. Polychaetes have only one synapomorphy, the nuchal organs (a pair of chemosensory structures on the postero-lateral margin of the prostomium), and the monophyly of polychaetes is still uncertain.

Polychaetes are abundant in most marine benthic environments, from intertidal to abyssal depths, from soft to hard substrates and from tropical to boreal regions.

GENERAL MORPHOLOGY

The body of polychaetes is divided into three parts: head, trunk, and pygidium (Fig. 11.1A). The head consists of prostomium, peristomium and in many cases, one or more cephalised anterior segments (Fig. 11.1B). The number, position and the shape of following appendages arising from the head region are important taxonomic characteristics: antenna (e), palps (tentacular crown and operculum, or buccal tentacles in some families), and eyes on prostomium, nuchal organs (caruncle in some families) from the posterior edge of prostomium, elongated cirri (usually called tentacular cirri) arising from cephalised segments. The definition of cephalised segments, however, is largely artificial, and differently defined between families.

In most polychaetes, the anterior part of the gut (pharynx or proboscis) is eversible and used for food capture or burrowing. This proboscis may have fleshy or chitinous papillae, and/or jaws. The trunk is the segmented region of the body between head and pygidium, usually consisting of a large number of almost uniform body segments. Body segments can sometimes be clearly divided into 2 or more body parts (e.g., thorax and abdomen) with different morphology (Fig. 11.1D). Even among the uniform body segments, the shape of parapodia and chaetae can change gradually or abruptly. Each parapodium is divided into notopodium and neuropodium, and may have dorsal cirri, ventral cirri, branchiae (gills) and other appendages (Fig. 11.1C). Chaetae are chitinous bristles; usually protruding from a pocket in the body

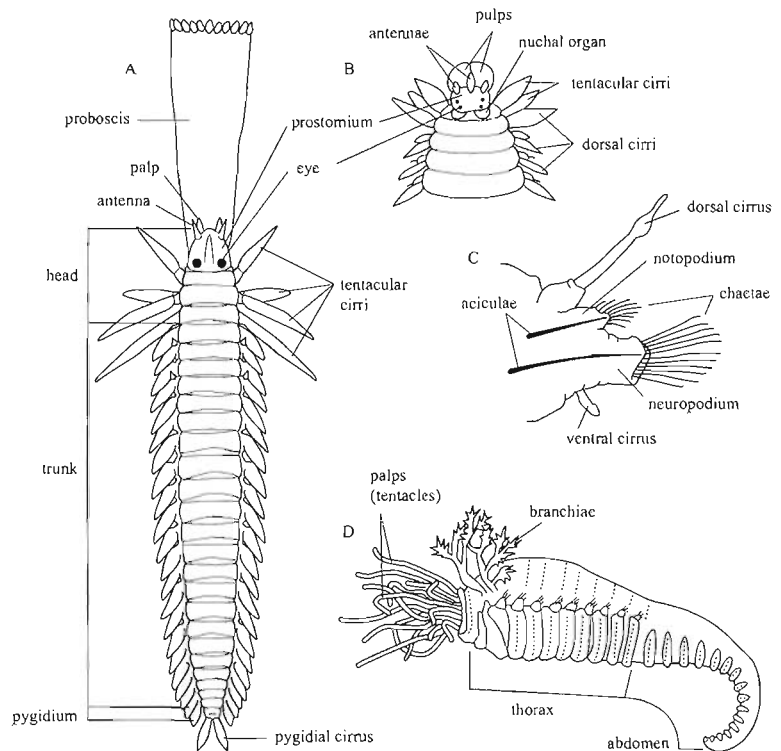


Fig. 11.1 **A** *Phyllodoce*, whole animal, dorsal view with everted proboscis; **B** *Syllis*, anterior part of body, dorsal view; **C** *Polynoides*, parapodium, anterior view; **D** *Terebellid*, whole animal, lateral view.

wall. Morphology of chaetae is quite various, and the shape, distribution and number of chaetae are used as taxonomic characteristics. The pygidium is the post-segmental terminal part of the body surrounding the anus, it lacks parapodia and chaetae but may have one or several pygidial cirri and/or papillae.

PREPARING SPECIMENS FOR IDENTIFICATION

Polychaete specimens should be anesthetized before fixation to ease future observation of the soft body parts. A freshwater solution of magnesium chloride $MgCl_2$ (see glossary of methods) works well for this purpose. Fix animals by placing them in 4% neutralized formalin in seawater (see glossary of methods). For long-term storage fixed material should be transferred to 70% ethanol.

The first step in the identification of polychaetes is to find a complete specimen, or at least a fragment including the head region. Polychaete specimens are frequently broken into pieces during sampling or fixation; and headless fragments are difficult or impossible to identify. Specimens in the best condition having all their head appendages (e.g., antennae, dorsal cirri, elytra) should be chosen. To identify the head region look for the mouth (not to be confused with the anus on the pygidium, although it can be difficult to distinguish which is which). In most cases, there are well-developed appendages on the head, and only simple structures on the posterior end. Most telling is the direction of the chaetae, as they are usually turned postero-laterally.

Most taxonomic characters needed to identify polychaetes are external; arrangements and morphology of appendages on the head region, and morphological features of parapodia and chaetae, and can be observed without dissection using a stereomicroscope (a compound light microscope may be used for smaller specimens). The number, shape and arrangement of proboscederal papillae and/or jaws are important taxonomic characters in certain groups of polychaetes, and for specimens with retracted proboscis this will require dissection. To examine a retracted proboscis, the anterior end of the specimen should be cut from the ventral side to avoid the destruction of the important characters on the head (Fig. 11.2A, 10.2B).

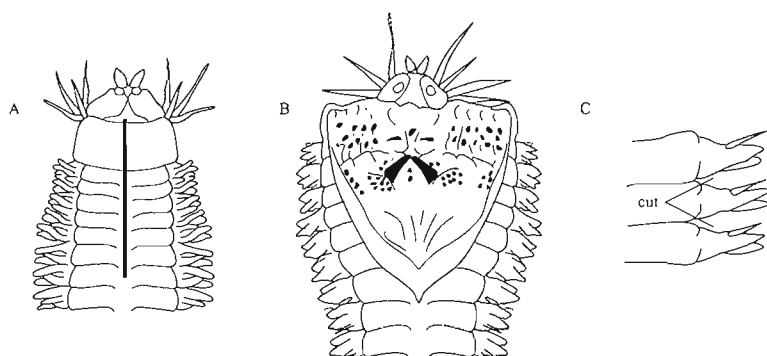


Fig. 11.2 Polychaete Dissection. A–C Nereididae. **A** head, ventral view, line indicates position of cut, **B** same, proboscis opened, **C** parapodia to be cut, dorsal view.

For specimens with a relatively short tough proboscis (e.g., Nereididae, Polynoidae) the ventral side of both body wall and proboscis need to be cut longitudinally for several segments. For specimens with a long fragile proboscis, the entire proboscis should be carefully removed from the body after cutting open the body wall, and dissected longitudinally in a small amount of glycerol on a glass slide (you can use a cover slip to keep the proboscis open as you work). After examination, the removed proboscis should be preserved along with the body (i.e. in the same vial).

The morphological features of parapodia should be examined carefully from both the anterior and posterior ends. Parapodia should be removed from the body using a scalpel or a small steel razor blade on a handle. To observe the difference of parapodial and chaetal features in different body regions, at least three parapodia from the anterior, middle and posterior part of the body should be examined. To determine the parapodia to be examined and to avoid damaged parapodia, the specimen should be carefully observed under a stereomicroscope. Parapodia should be cut along the anterior and posterior margin (Fig. 11.2C), and removed gently to avoid damage on parapodia and aciculae (if they are present). Removed parapodia should be transferred to a glass slide with glycerol, and examined under a compound microscope.

DIVERSITY

The classification of Polychaetes is continually changing. Traditionally, they have been classified into two groups: Errantia and Sedentaria, usually given the rank of subclass or order. This is currently considered to be an artificial grouping. Dales (1962) proposed a new classification, based on the structure of buccal organs and nephridia, dividing polychaetes into 14 groups (orders). Fauchald (1977) listed 17 orders largely similar to those of Dales, and Pettibone (1982) recognized 25 orders including orders for five 'archannelidan' families and new orders for Myzostomidae and Poeobiidae. Based on cladistic analysis using morphological characters Rouse and Fauchald (1997) proposed a new classification and that is the classification used here (Note the lack of taxonomic terms – subclass, order etc. as they are still being discussed):

Polychaeta are divided into Scolecida and Palpata.

Scolecida: Characterized by two apomorphies, presence of parapodia with similar rami, and the possession of two or more pairs of pygidial cirri. They are simple-bodied polychaetes, lacking antennae and palps (although some members of Paraonidae have a median antenna).

Palpata: Characterized by the presence of palps. Palpata is divided into Aciculata and Canalipalpa.

Aciculata. Characterized by the presence of aciculae, ventral sensory palps, prostomial antennae, dorsal cirri, ventral cirri, one pair of pygidial cirri, and segmental organs in most of segments. It is split into three groups:

Phyllodocida. Monophyly of Phyllodocida is indicated by the ventral position of sensory palps, the presence of anterior enlarged cirri (tentacular cirri), the loss of dorso-lateral folds, the presence of an axial muscular proboscis, and the presence of compound chaetae.

Amphinomida. Grouped together on the presence of calcareous chaetae. Comprise two families, Amphinomidae and Euphrosinidae.

Eunicida. Include polychaetes with a ventral muscularized pharynx with ventral mandibles and dorsal maxillae.

Canalipalpata. Characterized by an apomorphy of the presence of grooved palps and are split in to three groups:

Sabellida. Characterized by the presence of a limited prostomium fused to the peristomium, and the possible loss of peristomial lips.

Terebellida. Characterized by the presence of a first segment with no chaetae, a gular membrane, and a heart body. All members have either a pair, or multiple grooved palps.

Spionida. Characterized by the presence of a pair of peristomial grooved palps, nuchal organs forming posterior projections, and anterior excretory nephridia and posterior segmental organs for gamete release.

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Section 12 Cnidaria



(photo courtesy of T. Kato)

CHARACTERISTICS OF CNIDARIA

- 1) Cnidae
- 2) Radial or biradial symmetry
- 3) Single body opening
- 4) Diploblastic structure

INTRODUCTION

Members of the primarily marine phylum Cnidaria are diverse in morphology and life cycles. One of the two basic cnidarian body forms is a medusa (plural medusae), a deep or shallow circular dome with a central mouth beneath the dome, and tentacles fringing the edge of the dome. Typically living free in the sea (pelagic), its common name is jelly or jellyfish. The other cnidarian body form is a polyp, a cylinder with a mouth in the center of the disc at one end, and few to many tentacles surrounding the mouth. In a polyp that is part of a colony, the end opposite the mouth is attached to other members of the colony or to tissue connecting members of the colony; an example of colonial polyps are soft corals. In a polyp not part of a colony, the end opposite the mouth may be flat and attached to firm substrata, or rounded or pointed and burrowed into soft sediments; an example of a solitary polyp is a sea anemone.

Cnidarians are considered morphologically simple because of their radial symmetry (biradial in polyps such as sea anemones and corals), single body

opening, and body made of two tissue layers (thus they are diploblastic; most animals, including humans, are triploblastic, having three tissue layers). Yet cnidarians secrete the most complex intracellular structures known — cnidae, for which the phylum is named. All cnidarians possess the type of cnida (singular of cnidae) called nematocysts, microscopic stinging capsules with which cnidarians defend themselves and gather food. Nematocysts are a diagnostic feature of Cnidaria: if they are absent, the animal is not a cnidarian. However, their presence is not proof an organism is a cnidaria: nematocysts may be found in some predators of cnidarians. The cnidarian life cycle can also be complex: many species alternate between polyp and medusa forms, the polyps forming medusae through an asexual process, and the medusae forming eggs and sperm that ultimately develop into polyps. In species having only a medusa or only a polyp, that stage reproduces sexually, and may reproduce asexually as well. If asexually generated individuals remain attached to one another, the resulting group is considered a colony; if they separate physically, whether they remain beside one another or disperse, they are collectively termed a clone (i.e. each individual is not a clone but a clone-mate).

Medusae may be in the water over hard bottom shores or among seagrass blades, but most cnidarians in these habitats are polyps. Attached to stones or seagrass blades are commonly hydroids (class Hydrozoa) (some solitary, but most colonial) and sea anemones (class Anthozoa, order Actiniaria). Tube anemones (class Anthozoa, order Ceriantharia) may be burrowed into the sediment in which seagrass is rooted. Rare cnidarians on seagrass blades or algal thalli are stauromedusae (class Staurozoa) and medusae that crawl using their tentacles (class Hydrozoa, order Limnomedusae).

GENERAL MORPHOLOGY

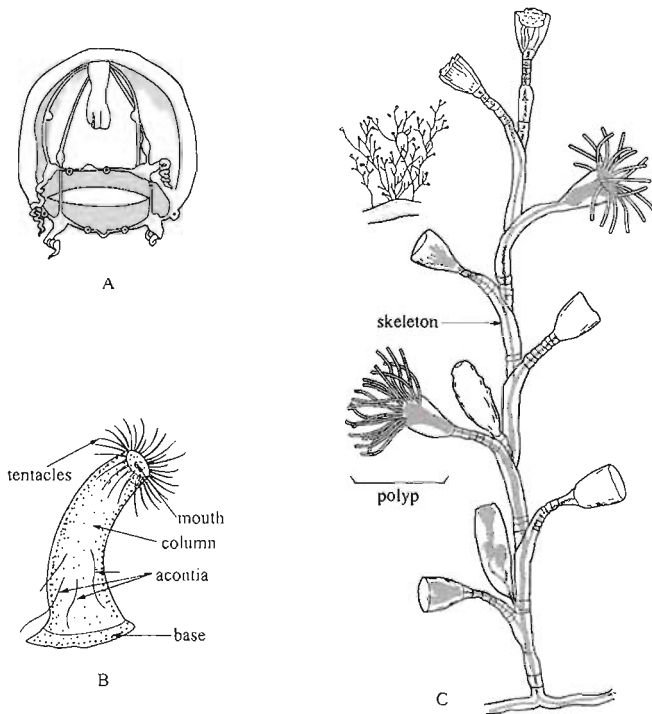


Fig. 12.1 A. A medusa of a hydrozoan (from Rufford, 1902). B. A sea anemone. Not all have acontia (nematocyst-studded threads emitted when the animal is disturbed) (From Verrill, 1928). C. Hydroid colonies. The larger, a close-up of part of the one at upper left, shows two polyps with tentacles expanded, four with tentacles retracted into the skeletal cup surrounding them, two empty cups, and two reproductive structures (from Rufford, 1902).

PREPARING SPECIMENS FOR IDENTIFICATION

Cnidarians should be relaxed before preservation. Common relaxants are magnesium salts, chloral hydrate, menthol, and nicotine (see glossary of methods). When the animal no longer responds to touch, pour off the water and add the preservative of choice. Preservation method depends on whether the organism is a medusa or polyp, whether it has a calcareous or chitinous skeleton, and the sorts of analyses planned. For example, if histology is used to identify a sea anemone, the polyp must be fixed (i.e. put into a liquid such as 10% formalin or Bonin's solution; after some days, it should be moved to a preservative such as alcohol). Tissue destined for molecular analysis should not be fixed but should be preserved in 95% ethanol; because molecular analysis requires little tissue, a piece of an individual or a colony may be removed for preservation in ethanol and the rest fixed or preserved in some other way. Because most taxonomy of scleractinian corals is based on the calcareous skeleton, all tissue may have to be removed to make an identification; typically this is done by placing the coral in a solution of household bleach. For cnidarians such as tube anemones and sea anemones, features of internal anatomy must be examined. For hydroids, form of the colony or type of medusa it produces may be essential, so identification of a single polyp may be impossible.

Nematocysts are useful or even necessary, in identifying sea anemone and some other cnidarian species. Nematocysts will not fire once the animal is preserved; keys are based on unfired ones. Nematocysts may be visualized by placing a very small pinch of tissue on a microscope slide in a drop of water, squashing it under a cover slip, and viewing with a compound microscope at a magnification of at least 400× and ideally 1000×.

DIVERSITY

Phylum Cnidaria is divided into five classes (two having been identified only rather recently), four with members that alternate between medusa and polyp stages. In these four classes, collectively termed Medusozoa, the duration of polyp and medusa stages may be unequal and the polyp and medusa stages may not be equally conspicuous; in some cases, one or the other stage is absent. The fifth class, Anthozoa, has only polyps.

Scyphozoa. “True” jellyfishes. Medusae larger and more conspicuous than polyps.

Cubozoa. “Box jellies.” Medusae larger and more conspicuous than polyps; some members can injure or even kill humans.

Staurozoa. Medusa shaped like an eight-pointed star attaches to a firm substratum; no polyp stage.

Hydrozoa. Medusae typically small, delicate, and short-lived. Polyps of many species colonial, form a skeleton, and have specialized types of polyps (i.e. some only feed, some only reproduce, some only protect the colony). Polyps with a chitinous skeleton are termed hydroids, those with a calcareous skeleton are termed hydrocorals. The Portuguese Man-o'-War, which is not a medusa but a pelagic colony mostly of specialized polyps, belongs to order Siphonophora. Members of order Chondrophora, also pelagic colonies, include the By-the-Wind Sailor.

Anthozoa. A polyp of subclass Octocorallia (= Alcyonaria) has eight tentacles, each with small side branches; almost all are colonial. Sea fans and sea whips (Order Gorgonacea) have stiff, flexible skeletons; a sea pen colony (Order Pennatulacea) has an internal strengthening rod throughout its entire length; blue corals (Order Helioporacea) have a calcareous skeleton. Some octocorals have polyps specialized for particular functions, such as pumping water through the colony. Polyps of subclass Hexacorallia (= Zoantharia), which are more variable morphologically than octocoral polyps, commonly have a multiple of six tentacles. Sea anemones (order Actiniaria), tube anemones (order Ceriantharia), and mushroom anemones (order Corallimorpharia) are exclusively solitary, black corals (order Antipatharia) exclusively colonial, and “true” corals (order Scleractinia) and zoanthids (order Zoanthidea) solitary or colonial.

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Section 13 Mollusca



(photo courtesy of T. Kato)

CHARACTERISTICS OF MOLLUSCA

- 1) Bilateral Symmetry
- 2) Mantle
- 3) Mantle Cavity (where respiration takes place and excretory and reproductive organs discharge)
- 4) Body divided into three regions: head, foot and visceral mass
- 5) Three coelomic spaces for kidney, heart and gonad
- 6) Radula (rasping organ)
- 7) Shell of calcium carbonate (can be reduced)

INTRODUCTION

Mollusks (or molluscs) are soft-bodied organisms, although most of them bear an outer shell of calcium carbonate. They constitute a very diverse group of about 72,000 living species occurring in marine, freshwater and terrestrial habitats. Sampling in seagrass, macroalgal and rocky shore communities will usually result in an abundant sample of mollusks, especially of snails, clams, and chitons.

GENERAL MORPHOLOGY AND DIVERSITY

The living mollusks are divided into seven classes:

Aplacophora: ~350 species in two subclasses: Chaetodermomorpha and Neomeniomorpha. They are exclusively marine; wormlike; instead of a shell

they have spicules or scales on the body surface. Most species are small and live in deep water.

Monoplacophora: ~25 species in a single order: Tryblidiida. They are exclusively marine with a limpet-like shell and are found in deep water.

Polyplacophora (chitons): ~1000 species in three orders: Lepidopleurida, Acanthochitonida and Ischnochitonida. They are exclusively marine with a shell of eight overlapping plates surrounded by a scaly or leathery girdle, very common in shore habitats with hard substrates.

Bivalvia: ~20,000 living species within four subclasses: Anomalodesmata, Heterodonta, Paleoheterodonta and Protobranchia. Found in both marine and freshwater their shell consists of two valves connected dorsally by a hinge and a flexible ligament, head not differentiated, lacking radula.

Cephalopoda (octopuses, squids, cuttlefish): ~900 living species in three subclasses: Nautiloidea, Ammonoidea and Coleoidea. They are exclusively marine and have elongated, tubular, tapering shells and live buried in soft bottoms.

Gastropoda (snails, slugs): ~70,000 living species in four traditional descriptive groups: Prosobranchia, Opisthobranchia, Gymnophora and Pulmonata. The taxonomy is currently under revision. The proposed classification divides the group in two subclasses, Eogastropoda (Patellogastropoda) and Orthogastropoda (Cocculiniformia, Vetigastropoda, Neritimorpha, Caenogastropoda, Heterobranchia). They are found in marine, freshwater, and terrestrial environments and usually have coiled shells (reduced or absent in slugs) and bilateral symmetry transformed through counterclockwise rotation of visceral mass and mantle cavity.

Scaphopoda (tusk shells): ~900 species in two orders Dentaliida and Gadilida, and eight families. They are exclusively marine and possess a well-developed head surrounded by arms and tentacles that bear suckers and hooks, jet propulsion provided by a funnel from the mantle cavity; shell modified, reduced or absent in most.

PREPARING SPECIMENS FOR IDENTIFICATION

Mollusks can be anaesthetized by placing them in the freezer until they are no longer responsive before fixing them in 70% ethanol or 4% neutralized formalin in seawater (see glossary of methods). For long-term storage they must be transferred to 70% ethanol, as formaldehyde will etch away the shell.

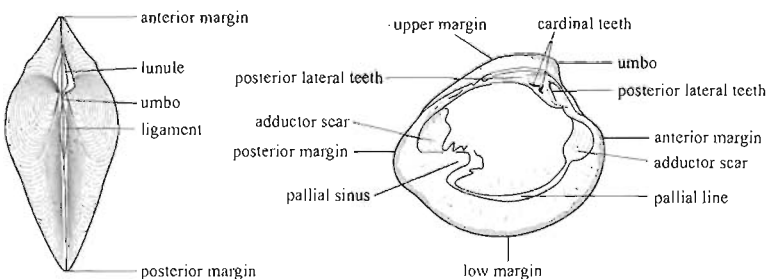


Fig. 13.1 Parts of the Bivalve Shell

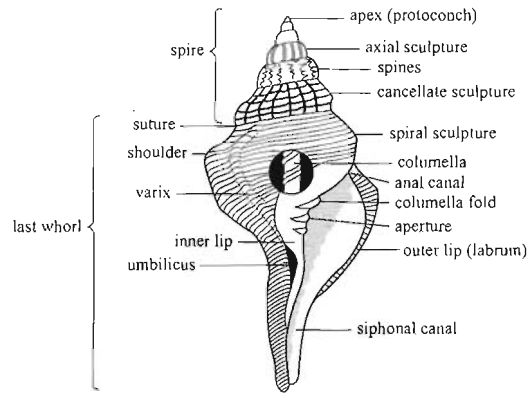


Fig. 13.2 Parts of the Gastropod Shell

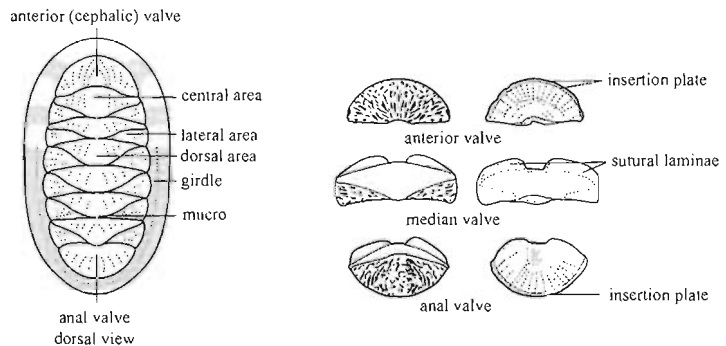


Fig. 13.3 Parts of the Polyplacophoran Shell

First, mollusks should be sorted into classes (chitons, gastropods, bivalves, and, if present, tusk shells and octopuses) before proceeding to identify them using appropriate illustrated local guidebooks and taxonomic keys. The shells of some gastropods may be overgrown and encrusted by barnacles, calcareous algae and other organisms, which obscure the sculpture and color pattern beneath. In such cases it is necessary to remove these encrustations using a pin or a metallic brush before attempting the identification.

With the exception of the Aplacophora, Cephalopoda and slugs, mollusk species can generally be identified down to family on the basis of their shell features (form, sculpture, hinge, color pattern, presence of nacre) based on photographs or drawings in a guide. Then the description in a taxonomic key of the likely families and genera of the specimen as well as the distribution ranges, habitat, etc. should be compared. To permit reasonable understanding of descriptions, it is necessary to become familiar with certain conchological (sea shell specific) terms (see accompanying sketches). An accurate identification at species level may be difficult for the beginner but as you practice you will improve your skills as a malacologist (a mollusk specialist).

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Section 14 Decapoda



(photo courtesy of T. Kato)

CHARACTERISTICS OF DECAPODA

- 1) Cephalothorax
- 2) First three thorax appendages function as mouthparts
- 3) Five pairs of walking appendages
- 4) Sometimes with chela (pincers)
- 5) Stalked compound eyes

INTRODUCTION

The decapod crustaceans are among the most familiar crustaceans and include shrimps, lobsters, crayfishes, and crab-like species. Their well-developed carapace, usually well calcified, is used as protection for the cephalothorax (head and body), including the branchial chamber where branchiae have developed as the major respiratory organs. Their name (Decapoda) means “ten legs”, referring to the five pairs of functional pereopods they use for walking or as defensive or feeding appendages (in some cases some of these legs end in a strong or weak pincer named “chela”). There are about 18,000 living species of decapods (Brusca and Brusca 2003) and they occur in all aquatic environments (fresh, brackish and marine waters) at all depths, and a few spend most of their lives on land. Many are pelagic (free-living in the water column), but many others

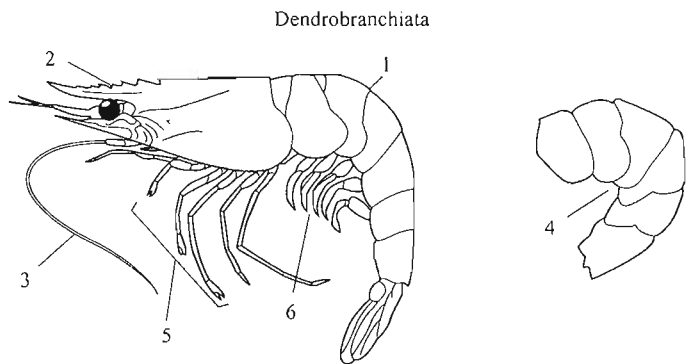
live on or close to the bottom (they are known as benthic species). Some even live within the sediment where they make burrows. Their feeding strategies include suspension-feeding, predation, herbivory, scavenging, among others. All species depend on water to lay their eggs. The first development stages are free living, pelagic larvae that pass through several stages before moulting to a small juvenile that will settle in its final environment. Some species have an abbreviated early development.

The nearshore, rocky and coral reefs environments are among the most diverse with respect to decapod crustaceans. These environments offer multiple microhabitats for protection (e.g. rock fissures, protected areas below rocks, dead and living coral, associated fauna like sponges, cirriped beds and mollusks, attached algae) and a large food supply. Rocky areas with strong wave action are usually less diverse than more-protected areas; small rocks and boulders also favor higher species richness than very large rocks as they offer a more protected area in term of refuges. Seagrass and algal beds offer good shelter for many species of decapod, although decapod species richness may be lower than in bare rocky shores where densities may be high. Species either live on the plant stems and leaves, or wander on or close to the bottom, under the plants. Many species of decapod living among corals, seagrass and algae are mimetic species that have the same or similar color (or even shape) as the substrate on which they live (i.e. the coral branch or the algal thallus). Some species of spider crabs also use algae, sponges or other material to decorate their carapace, making them hard to detect in their natural environment (Wicksten 1993).

GENERAL MORPHOLOGY AND DIVERSITY

Decapod crustaceans are divided into two major groups: Dendrobranchiata and Pleocyemata.

Group I. Dendrobranchiata (Fig. 14.1). Includes ~450 shrimp and like species. Most species live on the continental shelf and slope and in the

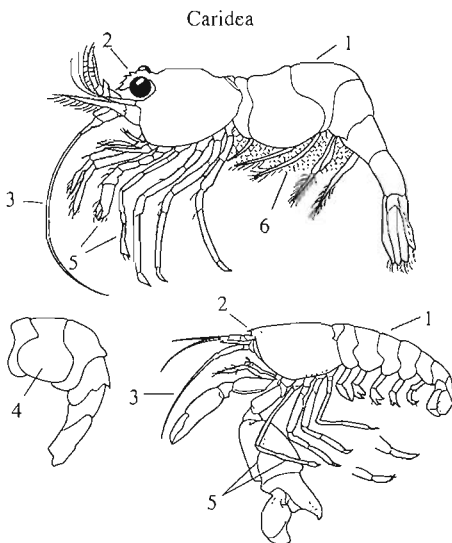


1. Body extended, abdomen long and robust.
2. Rostrum usually well developed, often spiny.
3. Antenna long, whip-like.
4. Lateral extension of abdominal segment 2 not overlapping segment 1.
5. Legs 1–3 ending in a small claw (pincer).
6. Females do not carry an egg mass.

Fig. 14.1 General Body Plan of Dendrobranchiata

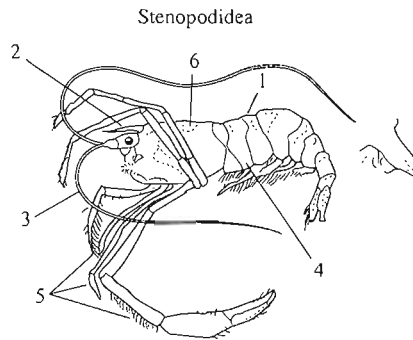
pelagic realm (“pelagic shrimps”). They are rarely found in the intertidal zone but many species come close to the coast during reproductive or recruitment periods. Many benthic species are of great economic importance and they are fished all over the world (Holthuis 1980). These decapods possess dendrobranchiate gills (two branches divided into multiple secondary branches), their body is fusiform, long, usually strong, with a well developed abdomen, often with a spiny rostrum (extension of the carapace), their first three pereopods end in a generally small chela (pincer). Contrary to all other shrimp-like decapods, females of this group do not brood their eggs. Fertilization of released eggs is external. Many of these animals are quite large, over 30cm long. Although some of the pelagic species might have a peculiar morphology due to adaptation to their environment (e.g. longer and slender appendages, spines, additional hairs), all dendrobranchiate shrimp look similar in shape.

Group 2. Pleocyemata Includes all the remaining decapods. Species included in this group never possess dendrobranchiate gills; they either have trichobranchiate gills (series of radiating unbranched tubular filaments) or phyllobranchiate gills (platelike or leaflike branches). The embryos are brooded by the female before they hatch (egg mass kept below the abdomen). This very large group of decapods includes several kinds of shrimps, the crabs, crayfish, lobsters, and many less familiar forms. Seven infraorders are currently recognized within the Pleocyemata but one (Astacidea) is almost exclusively from fresh water or deep marine habitat. The division into Natantia (swimming species) and Reptantia (walking species) has been abandoned as formal taxa, but is still found in many textbooks and guides.



1. Body extended, abdomen long and robust.
2. Rostrum usually well developed, often spiny.
3. Antenna long, whip-like.
4. Lateral extension of abdominal segment 2 overlapping segments 1 and 3.
5. Legs 1 and 2 ending in a claw (pincer), first often much larger than second.
6. Females carry egg masses.

Fig. 14.2 General Body Plan of Caridea



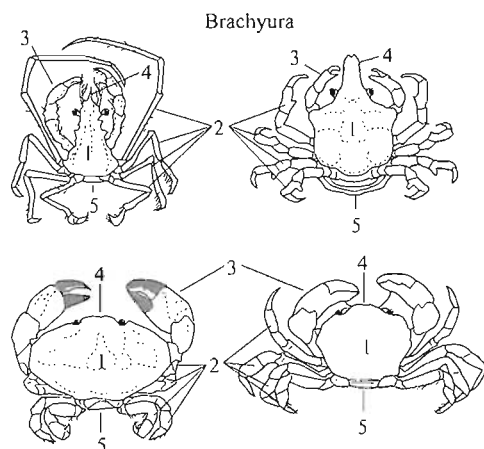
1. Body extended, abdomen long and robust.
2. Rostrum usually well developed, often spiny.
3. Antenna long, whip-like.
4. Lateral extension of abdominal segment 2 does not overlap segment 1.
5. Legs 1–3 ending in a claw (pincer), second much larger than the others.
6. Carapace often very spiny.

Fig. 14.3 General Body Plan of Stenopodidea

Caridea (Fig. 14.2). The caridean shrimps include over 2,500 living species and are mostly marine, although an important group of species (e.g. genera *Macrobrachium*, *Atya*, *Palaemonetes*) are found in fresh water (lakes, rivers) and in estuaries or brackish lagoons. The first 1 or 2 pairs of legs end in a chela (pincer) and vary considerably in size among species. The lateral part of the second abdominal segment overlaps both the first and third pleura. As in the case of the Dendrobranchiata shrimp, caridean shrimps are either pelagic or benthic. Deep-water species are often bright red, intertidal and shallow water species are usually brightly colored (particularly species associated with coral reefs) and estuarine species are generally whitish or transparent. Some species are important for fisheries (e.g. the Pandalid shrimps) (Hendrickx 1995). Many species are small and colorful and live in sponges, among corals, algae and seagrasses.

Stenopodidea (Fig. 14.3). With less than 30 species, this is a little known group of curious shrimps, all marine. The first three pairs of pereopods are chelate, and the third pair is larger than the others. The lateral portion of the second abdominal segment is not expanded as in carideans. Usually colorful, they are mostly tropical and benthic (coral reefs), although a few species are found in deep water. Shallow water species are often associated to other animals, like the cleaner shrimps of tropical fishes.

Brachyura (Fig. 14.4). This group, also known as the “true crabs”, contains over 10,000 species. The body is protected beneath a well-developed, calcified carapace. The carapace is flattened dorsoventrally, either smooth or ornamented with spines (lateral or dorsal), tubercles, or protuberances, and is oval, round, squarish, rectangular, triangular or polygonal in shape. The abdomen is symmetrical, flattened and reduced, and flexed beneath the thorax. Females keep the eggs between the flexed abdomen and the ventral part of the carapace until larvae are released. The first pair of legs, or chelipeds, ends in a chela (pincer) sometimes very much enlarged and adapted for defense, feeding or courtship. In many species chelipeds are asymmetrical and often larger in males than in females. Brachyuran crabs



1. Carapace flat, often bearing lateral or dorsal spines or tubercles.
2. Four walking legs.
3. First leg ending in claw (pincer).
4. Front with or without rostrum.
5. Abdomen flexed beneath carapace.

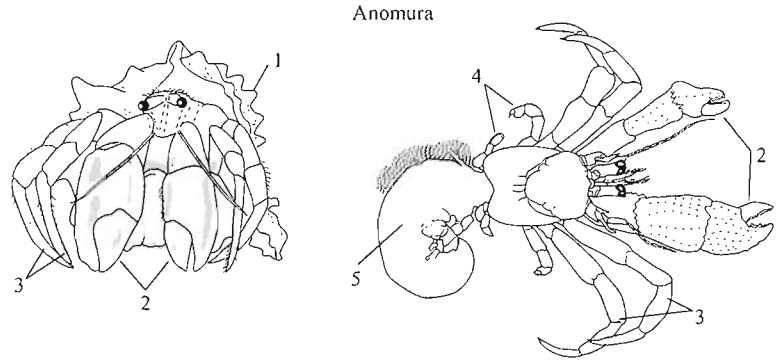
Fig. 14.4 General Body Plan of Brachyura

are mostly marine, but freshwater, amphibious and land crabs occur in the tropics.

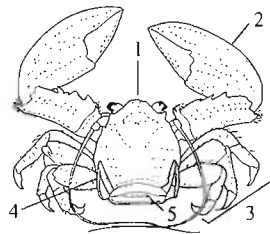
Anomura (Fig. 14.5). This group includes species with very different external aspects, like the hermit crabs, porcelain crabs, mole crabs, galatheid crabs, and king crabs. The latter two groups are only found in deep water while the others contain a various number of species occurring from the intertidal and shallow shelf water. Hermit crabs are typically found in empty marine snails shells in which they retract for protection; indeed, they possess a soft, asymmetrical twisted abdomen. Porcelain and mole crabs have a symmetrical, short, abdomen flexed beneath the thorax (as in true crabs); porcelain crabs are usually extremely flattened, very fragile (they tend to loose their legs when attacked or disturbed) and adapted for hiding in small, narrow crevices or below algae, shells or sponges. Mole crabs are oval to rectangular in shape and live in shallow water along sandy beaches, often in the surf zone where they hide in the sand. In all Anomura, the fifth pair of legs (and sometimes the fourth) are much reduced and are not used for walking.

Palinura (Fig. 14.6). Spiny lobsters and slipper lobsters are characterized by a strong, subcylindrical (spiny) or flattened (slipper) abdomen ending in a large tail fan. The chelation of the legs in Palinura varies but in spiny lobsters all five legs lack terminal claws (except in females). All species are marine and they are found in a variety of habitats throughout the tropics. There is a large fishery for lobsters around the world. Although spiny lobsters are commonly caught near shore, in rocky areas or in algal and seagrass beds, the other Palinura species live in much deeper water.

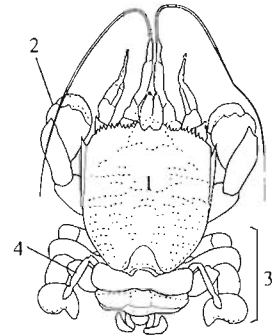
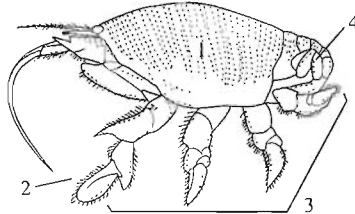
Thalassinidea (Fig. 14.7). Mud and ghost shrimps are particularly difficult to collect as many species make burrows that occasionally reach well over one meter deep. As their common name indicates, they look like small shrimp but they are also often confused with small lobsters. Many species



1. Generally living in an empty shell (marine snail shell).
2. First pair of legs ending in strong, symmetrical or asymmetrical claws.
3. Legs 2-3 used for walking.
4. Legs 4-5 reduced in size.
5. Abdomen soft, (generally) coiled and asymmetrical.



1. Body strongly depressed dorsoventrally, front slightly produced forward but without a true rostrum.
2. First pair of leg ending in flattened claw, opening in an horizontal plane.
3. Legs 2-4 used for walking; leg 5 reduced in size.
4. Abdomen partly flexed beneath carapace.

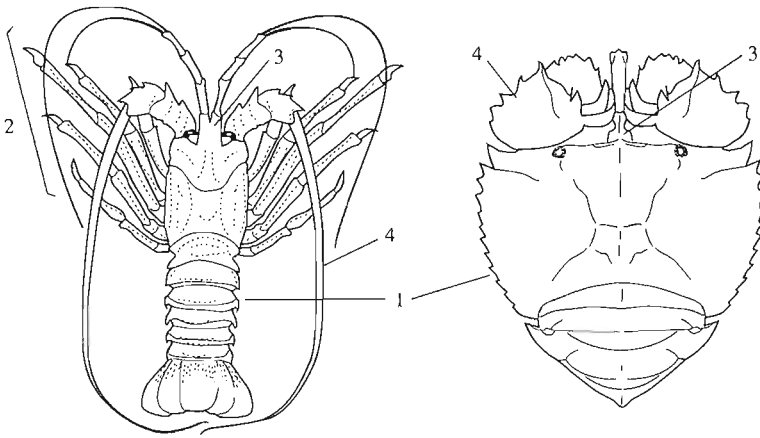


1. Body oval to subrectangular in shape, frontal margin with teeth or spines.
2. First pair of leg ending in a flat segment or in an imperfect (subchelate) claw, with moving finger closing vertically.
3. Legs 1-4 or 2-4 adapted for excavating in sand.
5. Leg 5 reduced in size.

Fig. 14.5 General Body Plan of Anomura

live in muddy substrate, but they are also occasionally found in rocky areas and among rubble, in deeper water, in muddy-sand or in mud. Their symmetrical abdomen is generally flattened dorsoventrally or subcylindrically, with a laterally compressed carapace; the first 2 pairs (or sometimes the first pair only) of legs are chelate, and the first pair is usually much larger than the second. Mud shrimps are very characteristic of tidal flats, generally

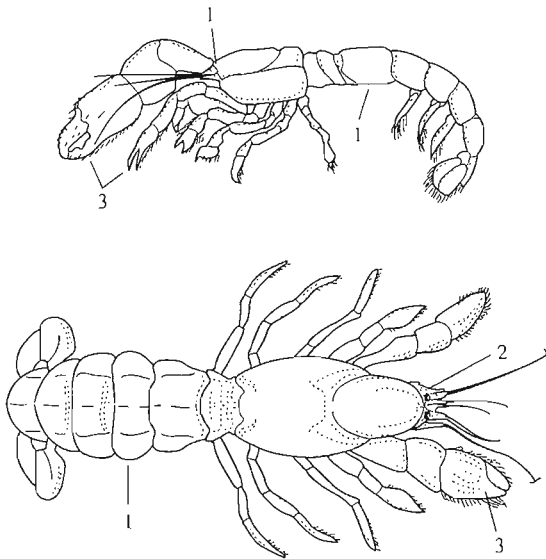
Palinura



1. Body extended, abdomen long and robust, cylindrical or dorsoventrally flattened.
2. Five strong, similarly shaped walking legs, not ending in a pincer (except for a small pincer on leg 5 in females).
3. No conspicuous rostrum.
4. Antenna long, wipe-like and spiny (spiny lobsters) or flattened (slipper lobsters).

Fig. 14.6 General Body Plan of Palinura

Thalassinidea



1. Body extended, usually depressed dorsoventrally with reduced rostrum and carapace weakly calcified.
2. Rostrum spiny and carapace well calcified.
3. Legs 1–2 (or only 1) ending in a claw, the first usually much stronger, long and laterally compressed.

Fig. 14.7 General Body Plan of Thalassinidea

within or close to estuaries and lagoons where they burrow holes in the soft sediment and form large colonies.

PREPARING SPECIMENS FOR IDENTIFICATION

Examination of live specimens is not always easy as they move quickly, especially when light is used. A mild anaesthetic can be used to slow them down (a few drops of clove oil or a 1% seawater solution of propylene phenoxetyl). Care should be taken with large crabs as their powerful claws can cut or break a finger. Decapod specimens collected in the field should be fixed shortly after collection. Preferably they should be placed in a freezer in a plastic container, which allows a painless death before being fixed with a mild solution (4%) of neutralized formalin in seawater (see glossary of methods). After a few days, specimens must be washed with plenty of tap water (leave the material in tap water overnight after washing it two to three times) and preserved in a 70% ethanol solution and examined under a dissecting microscope. Usually a macroscopic examination is sufficient to recognize specimens to species level.

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Part IV

Specific Details for Working with the Secondary Target Groups

Section 15 Porifera



(photo courtesy of C. Diaz)

CHARACTERISTICS OF PORIFERA

- 1) Composed of outer pinacoderm, inner choanoderm and intermediate mesohyl
- 2) Amorphous to regularly branching growth
- 3) Internal water channel system
- 4) Internal skeletal elements of spicules and spongin fibres
- 5) Sessile lifestyle

INTRODUCTION

Sponges are motionless, aquatic, benthic organisms with a unique body plan within the Animal kingdom. A sponge specimen may look like an amorphous mass, a branching tree, a perfect sphere, or a thin skin. Its body is formed by aggregations of cells differentiated into two layers of ill-defined tissues (the pinacoderm and the choanoderm). These flank a middle “matrix-like” layer (the mesohyl) that contains various cell types (reproductive, skeleton-secreting, undifferentiated, or specialized cells, and bacteria), and the skeletal elements (spicules and/or collagen fibers). These basic three layers line up around a system of pores, canals, water-pumping chambers (covered by specialized flagellated collared-cells), and oscules (excurrent openings), making the sponge the most efficient water-filtering organism in the sea. Sponges are present in all aquatic environments. In the ocean they occur from low to sub-tidal depths, on rocky outcrops and crevices where macroalgal communities splurge, to the deepest parts of the seas. In the rocky seashore sponges are usually small, while in seagrass beds sponges can be small thin crusts overgrowing seagrass blades to stand alone organisms as large as truck tires.

GENERAL MORPHOLOGY

The sponge body shows distinct features such as: shape, color, oscule morphology, surface features, and consistency, which may characterize a

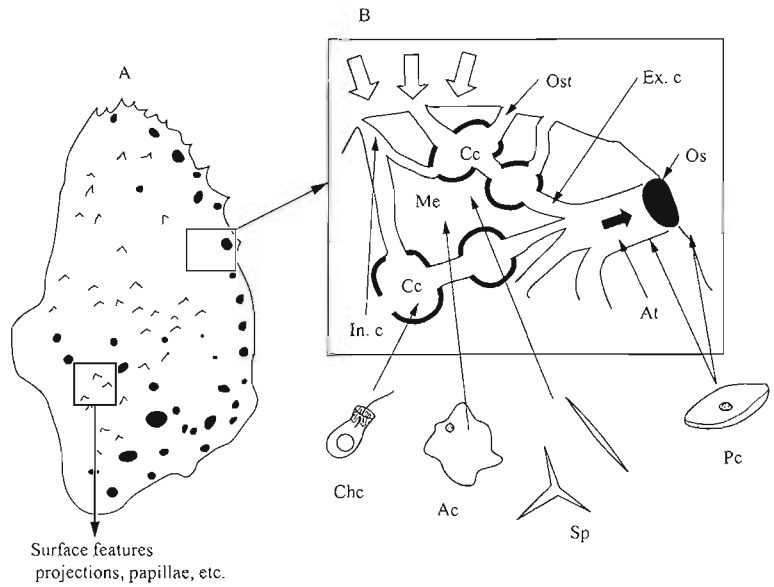


Fig. 15.1 The Sponge Body Plan

A. A schematic drawing of *Amphimedon compressa* from the Caribbean showing a massive flabellate body riddled by oscules (large excurrent openings), and few surface features. Scale 1 cm = 1 cm. **B.** Schematic drawing of a hypothetical cross section through the sponge showing the direction of water filtration: ostia (Ost) – incurrent canals (In. c) – choanocyte chambers (Cc) – excurrent canals (Ex. c) – atrium (At) – oscule (Os), major cell types and their location: choanocyte cells (Chc) lining the choanocyte chambers, pinacocyte cells (Pc) cover the internal and external sponge surfaces, amebocyte cells (Ac) on the mesohyl (Me), and spicules (Sp) secreted from the mesohyl. Arrows indicate where the water enters (open) and where it leaves the sponge body (black).

particular species (Fig. 15.1A). The surface of the sponge reflects the morphology of the skeleton under it. Depending on how the sponge skeleton reaches to the surface a species may produce a different texture effect on the sponge “skin”. Projections shaped like small cones (conulose), spikes, spatula, or with irregular shapes may characterize members of a species or even a genus. Smooth surfaces usually reflect the existence of a specialized skeleton layered tangentially to the sponge surface.

Sponges do not have a mouth or a digestive tract. Ambient water enters their body through small pores (ostia) and follows through incurrent canals that reach flagellated “water-pumping chambers” (choanocyte chambers). These chambers open into excurrent canals that converge into a larger cavity (atrium) that reaches the outside through the oscule (Fig. 15.1B). These water-filtration systems present various grades of complexity, ranging from the simplest body with a single central cavity lined by choanocytes (Ascon) to those with choanocytes restricted to small chambers and with simple canal systems (Sycon) to those with complex and numerous subdivisions of the internal canals (Leucon) such as the one represented in Figure 15.1.

Sponges possess an internal skeleton, which provides support for growth. The skeleton is composed of one or more of the following elements: spicules

(mineral secretions of silica or calcium carbonate), fibrillar collagen, and a type of collagen exclusive of sponges; spongin, which is found in the form of fibers, spicules, or plates. Spicules occur in many different sizes and shapes. Large spicules (the megascleres) usually form the main skeletal framework, and are classified by the number of axes (monaxon, triaxon, tetraaxon), the number of rays (monoactineal = one ray, to hexactineal = six rays), and other special features (spines, rounded ends etc.). Small spicules (the microscleres) usually fill in the main skeleton, or are located in particular areas of the body (surface). Microscleres come in a great array of shapes (stars, chela etc.) and are an important classification tool.

PREPARING SPECIMENS FOR IDENTIFICATION

When collecting voucher specimens of sponges or sponges within the 25cm quadrat the specimen should be carefully studied. Characters such as shape, size, color (external and internal), oscule morphology (shape, distribution, and abundance), texture (surface features), and consistency (hard, soft, glassy, etc.), descriptions of its habitat, substrate, depth, and observations about its neighbors may result useful to characterize a species. Sponges should be removed from the substrate where they grow using a knife, or another cutting tool, and preferably wearing diving gloves to avoid contact with toxic chemicals or sharp piercing spicules. For identification purposes (i.e. in the 1m quadrat) only a photograph and small portion of a sponge needs to be taken, leaving the major functional body parts (oscula, peduncle, outer surface) intact to allow the recovery of the sampled specimen. The sample should contain a portion of the external and the internal body parts since the skeletal structure of those parts might be very different.

For histological purposes a small piece of the sponge must be placed in a fixative solution such as 4% neutralized formalin-seawater solution, Bouins' fixative, or Glutaraldehyde 2% (in Cacodylate or related buffer). The sponge must be transferred within a week to a 70% ethanol seawater solution. Never place a calcareous sponge into a formalin solution since the calcium carbonate will be dissolved. If the sponge is only being preserved for skeletal studies then it can be directly placed into 80 to 90% alcohol. For DNA studies a small sponge piece (2 g) can be placed in >90% Ethanol or chopped into less than 1 mm pieces and placed into a 1.5 cc tube filled with Silica Gel (electrophoretic grade) for later DNA extraction and analyses.

The first question to answer is what type of skeletal elements the sponge has: calcareous spicules, spongin fibers assembled into fibers and/ or reticulations, and/or siliceous spicules.

A rapid acid test (see glossary of methods) will allow you to identify a calcareous spicule, which could belong to a calcareous sponge (di-, tri-, and tetractineal in shape), or another animal phylum such as didemnid ascidian (round spined spicules), or a soft coral (long spined spicules). If cleaner or permanent preparations are required it is more convenient but more tedious to use acid digestion (see glossary of methods).

The next step is to classify the sponge skeleton. For this it is necessary to make thin sections of preserved material (Dr. Klaus Ruetzler, per.comm, see glossary of methods for an explanation of the technique using both wet and dry specimens). Other methods are used for histological studies. Sections of paraffin or epoxy embedded tissues are made by hand, microtome, or with cutting and polishing techniques (used in geology and paleontology). These

methods can be found in histological or paleontology literature, and should be discussed with sponge experts.

A guide to Porifera classification at levels higher than species was put together through the collaboration of more than twenty sponge experts (Systema Porifera, Hooper & Van Soest eds.) in 2002. It provides a practical guide with clear explanations and illustrations of the morphological characters used for the taxonomy and systematics of each higher taxon within the sponges. Within each sponge taxa, characters such as external morphology, skeletal elements (shape and size) and skeletal arrangement have a distinct diagnostic value. For example, species differentiations within a genus rely mostly on external characters (shape, surface features, etc.), spicule sizes, or special spicule features. Families and orders are distinguished on the basis of skeletal framework architecture, spicule types, and choanocyte chamber morphology. When the skeletal elements are absent, characters such as cell types, reproductive structures, and sometimes chemistry (secondary metabolites) can aid in the differentiation of certain sponge groups. Genetic and molecular information (protein electrophoresis, DNA sequencing) are proving essential to discern the affinities on groups with low to none morphologic diagnostic characters. They also provide means to study population exchanges and the phylogenetic relationships within this animal group.

DIVERSITY

Sponges inhabit all aquatic habitats, and have been an important faunal component of marine systems since the early Cambrian era (560 mya). Approximately, 10,000 species have been described; however, their diversity is believed to reach at least 15,000 species. Extant sponges are currently classified into three major classes: Hexactinellida, Demospongiae, and Calcarea.

Hexactinellida or “glass sponges” (approximately 500 species) possess triaxonic hexactineal spicules (six rays), which usually form a tight mesh giving the sponge a glassy consistency. The pinacoderm and choanoderm in Hexactinellida are syncytial (cells are fused), and with respect to body complexity they are either Sycon or Leucon. These sponges occur only in deep marine waters (200–5000 m) and can reach massive sizes.

Calcarea (approximately 500 species) possess spicules of calcium carbonate, usually a combination of monoaxonic, triaxonic and tetraxonic forms in one or various size classes. They are exclusively marine and present the three types of structural organization (Ascon, Sycon, and Leucon). Specimens are usually small (a few mm, to 10–15 cm in length) and usually occur at shallow depths.

Demospongiae (approximately 10,000 species described) include 90% of the extant sponge diversity, inhabiting all aquatic environments. Their skeleton is made up of a combination of fibrillar collagen, siliceous spicules and/or the spongin fibers, spicules, or plates.

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Section 16 Amphipoda



(photo courtesy of M. Shimomura)

CHARACTERISTICS OF AMPHIPODA

- 1) Small (usually less than 1cm)
- 2) Laterally compressed
- 3) First four pairs of legs directed forward, last three pairs directed backward
- 4) One head, seven thoracic, three pleonal, and three urosomal segments
- 5) Three pairs of uropods on the urosome

INTRODUCTION

Amphipods are very common small crustaceans living mainly in marine and freshwater environments, although some live their entire lives in supralittoral and terrestrial domains. More than 6000 species are described from the marine waters, and they are considered to be one of the more important groups of invertebrate animals in the benthic realm (although almost 200 species live planktonically).

MORPHOLOGY

These crustaceans are distinguished very easily by the fact that the first four pairs of legs are directed forward, unlike the last three, which are directed backward; that is why they are named “amphipods”; amphi-both, pods-legs, in Latin. Amphipods usually are small (<1cm in length, although much larger species exist) and are laterally compressed. The head is followed by seven pereon (thoracic), three pleonal, and three urosomal segments. As in all crustaceans, one appendage from each segment is articulated. On the pereon are two pairs of gnathopods and five pairs of pereopods. On the pleon are three pairs of pleopods. On the urosome three pairs of uropods are always present. The telson is a very small additional piece at the posterior end of the animal.

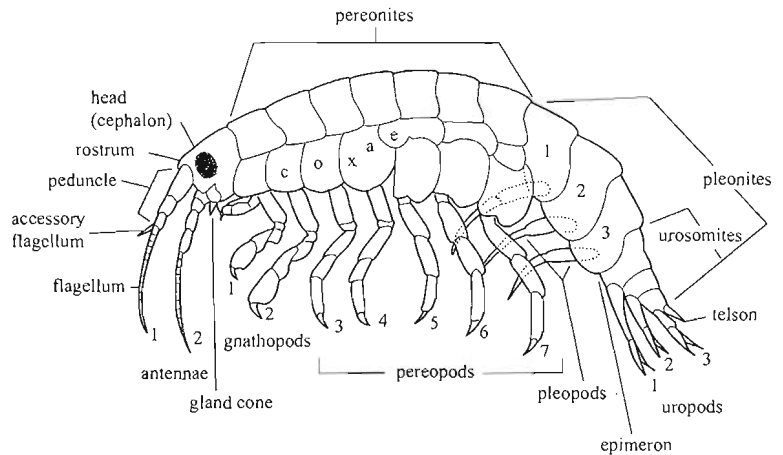


Fig. 16.1 General Amphipod Body Plan

As they are peracaridean crustaceans they have no free larval development, the eggs, embryos and juveniles are carried in the female's marsupium or brood chamber, which is ventrally situated, and consists of the overlap of the female's oostegites. The paired sides of the marsupium usually arise from coxae two through six of the adult females.

PREPARING SPECIMENS FOR IDENTIFICATION

Lay the amphipod on an excavated slide with one to three drops of glycerin (dilute the glycerin with water when handling small specimens). Dissections should be carried out with a stereomicroscope (using a reflected light, or an optic fiber lamp), although the dissected pieces should be transferred to a compound microscope.

The small size of the amphipods makes dissection difficult for the beginner but not impossible, as with all new things practice is important. Start with the dissection of any of the legs, which are usually the largest pieces, and move on to the mouthparts, which are very important for identification.

The first step is to identify which suborder the specimen belongs to: Gammaridean, Hyperiidean or a Caprellidean (Ingolfiellidean are very rare and are unlikely to be found in the near shore zone). The second and most important characters to be checked are the type of accessory flagellum, when present; the type of eye; if the coxae are touching each other or not, the presence of dorsal teeth on the pereon or pleon; the morphology of uropod three; the type of gnathopods and legs; and the morphology of the mandibular palp, if present.

DIVERSITY

Amphipods are separated taxonomically into the planktonic amphipods (Hyperiidea) and benthic amphipods (Gammaridean, Caprellidean, and Ingolfiellidean).

Gammaridean amphipods have 124 families, all with basic morphology. They are mainly benthic animals with well-developed bodies and maxillipeds bearing palps, although some do lack eyes.

Caprellidea (ghost or skeleton shrimp) consist of 8 families. Their body is elongated, not laterally compressed, and with a small abdomen. Caprellids hold on to the substratum (seaweeds, hydrozoans, bryozoans) to catch prey with well-developed sub-chelate claws on their thoracic appendages.

Hyperidea have 2 Infraorders, and 21 families. They are exclusively planktonic and have large eyes and well developed pleopods. The maxilliped, which is reduced, never has palps. Some have a lanceolate body, but the large head easily identifies them as a planktonic amphipod.

Ingolfiellidea is a very rare and small group of interstitial or marine animals that lives in deep waters and belongs to a single family.

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Section 17 Isopoda



(photo courtesy of M. Shimomura)

CHARACTERISTICS OF ISOPODA

- 1) Body divided into three regions: *head* of fused segments; *pereon* of seven segments, usually free; and *abdomen* of five segments, sometimes fused, plus pleotelson (fused last segment and telson)
- 2) *Head* with two pairs of antennae, mandible, two pairs of maxilla, and maxillipeds
- 3) *Pereon* with seven pairs of pereopods (often but not always similar walking legs), each of only one branch
- 4) *Abdomen* with five pairs of pleopods plus one pair of uropods attached to pleotelson
- 5) Females carrying eggs, embryos and juveniles in a marsupium sitting under the pereon and derived from branches of the pereopods (as in all peracarid crustaceans)
- 6) Males with pair of stylets on inner edges of second pleopods
- 7) Juveniles without last pairs of pereopods (six pairs only)

INTRODUCTION

The Isopoda are one of the orders of peracarid crustaceans, that is, those that brood their young in a marsupium under the body. They are uniquely defined by the combination of one pair of uropods attached to the pleotelson and pereopods of only one branch. The order is the most morphologically diverse of all peracarids, including flattened forms with similar legs, cylindrical bodies with legs very different from each other, and sexually dimorphic parasitic forms.

Most of these forms are found in the nearshore rocky environment, some specialising as burrowers in sand or mud and others as inhabitants of algae or seagrasses.

PREPARING SPECIMENS FOR IDENTIFICATION

A stereomicroscope with a magnification of at least 25 times is essential as is a good light source (fibre-optic). Reflected light is good for seeing sculpture but transmitted light is better for counting setae although it is a

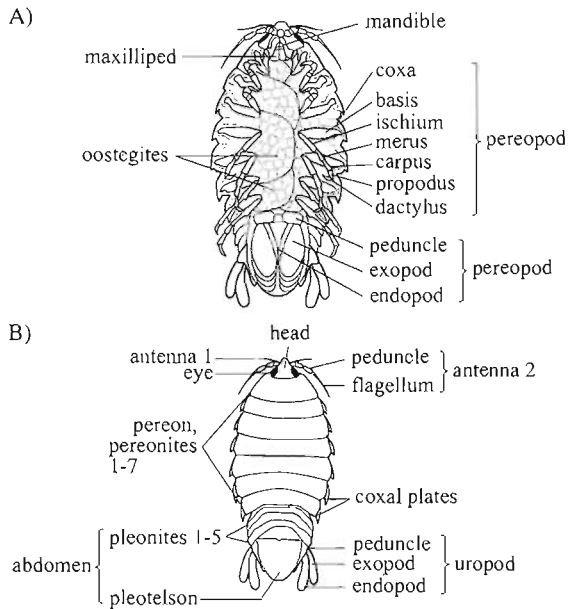


Fig. 17.1 Cirolanid Isopod

A) Ventral and B) Dorsal Views (Kensley, 1978, relabelled)

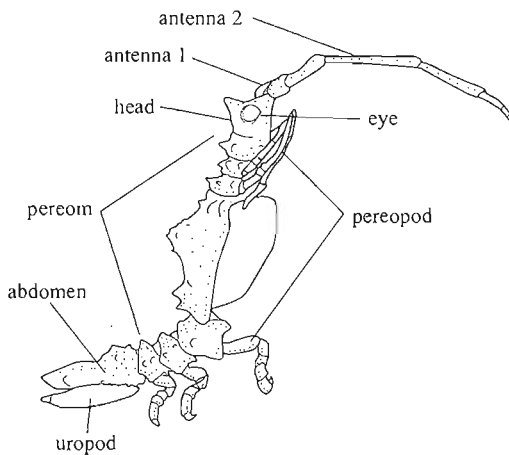


Fig. 17.2 Arcturid Isopod, Lateral View (Poore, 2001b)

matter of preference. Examination of most characters is facilitated when the specimen is immersed in alcohol (70% is standard) and manipulated by fine forceps and a sharp dissecting needle.

Isopods can be identified to species on the basis of external morphological features of the body and limbs. Shapes and sculpture of body parts, counts of the number of segments or articles, the degree of fusion between them, and the number of setae or spines are critical features.

Usually specimens can be identified to family without the need to dissect limbs or mouthparts from the body. The first thing to look at is the abdomen

and arrangement of uropods, whether or not the uropods project backwards, are in contact with the pleopods, or enclose the pleopods. This will provide a quick assessment of the suborder. The degree of fusion of pleonites, how many segments are visible dorsally is often a key to families. Next most important is the relative shapes of the pereopods, whether or not they are similar, or which anterior ones are setose or hooked will help define the family. A compound microscope can be used to differentiate species by examining setation or mouthparts after they have been dissected from the body. To dissect an isopod place the specimen in glycerol and dissect parts off with a needle; then transfer the dissected limbs to glycerol on a microslide and cover with a coverslip.

DIVERSITY

The order Isopoda is classified into seven suborders not all of which are found in macroalgal and seagrass communities. Several families are micropredators or parasites on fishes or decapods and rarely encountered freeliving in these environments. The suborder *Oniscidea*, with many families are the most familiar isopods, slaters and woodlice, but are entirely terrestrial and have few representatives associated with marine habitats, and then only high on the shore. The truly marine and freeliving suborders likely to be found in coastal seagrass and macroalgae are:

Cymothoida. Includes three freeliving superfamilies:

Cirolanoidea, with one family *Cirolanidae*, are the most commonly seen typically flattened sea lice.

Anthuroidea, with six families, are, elongated isopods with uropodal exopods standing erect over the pleotelson.

Cymothoida, including the families (among others) *Aegidae* and *Corallanidae*, usually having free pleonites and hooked pereopods; and *Gnathiidae*, of which the males have grossly enlarged mandibles. All are micropredators of fishes spending much of their time away from their hosts.

Limnoriidea. Including families Limnoriidae, Keuphyliidae and Hadromastacidae, all with hooked tips on the uropods. Limnoriids bore tunnels into macroalgae and wood.

Sphaeromatidea. Including two superfamilies:

Sphaeromatoidea (Sphaeromatidae and other minor families), the pill bugs, with segments of the abdomen fused into two units

Seroloidea (Serolidae and other minor families), extremely flattened isopods with extended coxal plates and a usually enlarged first pereopod.

Valvifera. Including families Idoteidae, Holognathidae, Arcturidae and other families – isopods in which the uropods fold under the abdomen to enclose the pleopods in a branchial chamber

Asellota. Including Munnidae, Janiridae, Jaeropsididae and Paramunnidae in shallow water plus many other families confined to the deep sea, small isopods with the first and second pleopods modified as an operculum over the remaining pleopods

Many papers, websites and books on isopods recognise the now obsolete traditional isopod suborder “Flabellifera”, a catchall grouping of the families now included in Cymothoida, Limnoriidea and Sphaeromatidea. The keys to families of Flabellifera in these older works are still useful for practical purposes.

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Section 18 Meiobenthos



(photo courtesy of Y. Shirayama)

INTRODUCTION

Meiobenthos is not a taxonomic group but rather a size class of the benthic community (Mare 1942), which is smaller than 1–0.5mm and larger than 32–63 μ m. In NaGISA, it is defined as those organisms that pass through the 0.5mm mesh sieve and are retained by the 63 μ m mesh sieve. Meiobenthos consists of Metazoa and Protozoa. The dominant metazoan taxa are Nematoda and harpacticoid Copepoda. Usually the former accounts for about 80% of total metazoan meiobenthos, and the latter for about 10%. Other meiobenthic metazoans include: Kinorhyncha, Gastrotricha, Loricifera, Tardigrada, Ostracoda, Turbellaria, Nemertenea (Fig. 17.1). Among protozoans, Foraminifera and Ciliata are the most abundant.

Along with the permanent meiofauna listed, juveniles of macrofauna (e.g. Polychaeta) are also found in meiobenthic samples, and are considered temporary meiofauna. Meiobenthos is abundant in both rocky nearshore habitats and seagrass beds. The number of individuals living per m² can often be as high as 10⁷.

MORPHOLOGY AND DIVERSITY

The only unification in a meiofauna sample is size and so there is a considerable amount of morphological diversity found within a meiofaunal sample. Foraminifera is the most abundant protozoan meiofauna. The species with calcareous shells are easier to identify than those that do not create a test or use sedimentary grains to do so. Nematoda is the most dominant metazoan phylum and specimens with their thread-like appearance under dissecting microscope are easily distinguishable. Nematodes with unique in



Fig. 18.1 Representative Species of Meiofaunal Phyla

1: Ectoprocta; 2: Annelida; 3: Chordata; 4: Mollusca; 5: Echinodermata;
6: Sipunculida; 7: Nematoda; 8: Priapulida; 9: Loricifera; 10: Kinorhyncha;
11: Turbellaria; 12: Tardigrada; 13: Brachiopoda; 14: Arthropoda;
15: Rotifera; 16: Gnathostomulida; 17: Gastrotricha; 18: Cnidaria; 19: Ciliata;
20: Nemertinea; 21: Entoprocta; 22: Foraminifera (Courtesy of R.P. Higgins)

morphology (for example, Desmoscolecidae) can be keyed out to family level very easily. Harpacticoid Copepoda are also easily identified if it is in its copepodite stage, but it is difficult to distinguish nauplii of this group from other crustaceans such as; Ostracoda, Tanaidacea, Cumacea and Amphipoda, which are also abundant in most meiofaunal samples. Kinorhyncha are easily separated from their groups of animals with segmented bodies by the lack of segmented legs and their many anterior scapids. Juvenile of or small species of Annelida (Polychaeta and Oligochaeta), Mollusca (Gastropoda, Bivalvia, Solenogastres etc.), Echinodermata (mainly Ophiuroidea), and Sipunculida are also abundant in meiobenthos. Other phyla such as Loricifera, Priapulida, Nemertenea, Entoprocta, Ectoprocta, Brachiopoda, Rotifera, and Urochordata are rarely found. It is difficult to identify especially soft-bodied meiofauna such as Ciliata, Turbellaria, Gastrotricha and Gnathostomulida from formalin-fixed material, sometimes even to the phylum level. It is, therefore, important to work with live material as much as possible.

PREPARING SPECIMENS FOR IDENTIFICATION

Meiofauna can be stored in 10% neutralized formalin in seawater (see glossary of methods) until sorted, unless taxonomic voucher specimens are to be prepared, which requires fresh specimens to be anesthetized using $MgCl_2$ or *l*-Menthol (Higgins and Thiel 1988) before sorting them to phylum level under a stereomicroscope with a needle or "Irwin's loop"

(Higgins and Thiel 1988). Fixing methods differ between taxa: Nematoda can be preserved in 5% neutralized formalin in seawater; taxa with calcareous skeletons (e.g. foraminifers, crustaceans, kinorhynchans) should be preserved in 70% ethanol in distilled water or 20% ethylene glycol in distilled water.

A compound microscope is necessary to make detailed observations of specimens. To prepare microscopic slides preserved specimens should be transferred to a solution of 10% glycerol with thymol (several pieces per 500ml glycerol), 30% Ethanol in distilled water and placed in a drying oven at 70°C to evaporate the ethanol and water. Using a Cobb's aluminum frame or H-S slide (Shirayama *et al.* 1993) will allow the specimen to be observed from both ventral and dorsal (or both left and right) sides.

Drawings and photos are invaluable for identification but should only be made after observing all samples and selecting the specimen in the best condition to make the most accurate representation.

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Part V
Beyond NaGISA;
Further Information and
Optional Protocols

Section 19

Practical Coastal Physical Oceanography for Ecologists

THE COASTAL ENVIRONMENT

Every species, population or community is to different degrees, related to its environment. Coastal environments are normally a very physically dynamic, and significant changes occur over a very short time period e.g. the changes in water level induced by the tides or, in a shorter period, by the individual waves. The coast acts as a barrier to circulating water masses. In open sea (beyond the continental shelf), water circulates in three-dimensional space with few interactions with its boundaries (sea floor and land masses). On the coast, these boundaries are much closer and thus more interconnected to the processes, which are turbulent and complex.

The coastal environment has characteristics that make it different from the open sea. The seafloor at very shallow depths induces frictional stress over the water masses, which results in an increase of bottom sediment transport. The presence of the coast generates convergences and divergences: the water currents accelerate; waves deflect, reflect and refract. If the wind guides the water offshore, large seasonal upwelling systems can result. The tides can be amplified or substantially modified by the morphology of the coast (estuaries, embayments, etc.) and the seafloor. The terrestrial influence is of great importance. Freshwater runoff can locally modify the salinity; large rivers add huge amounts of fine sediments; nutrients from land affect the productivity of the system. Even the climate in the coastal areas is influenced by the presence of landmasses. And finally, the coastal environments are prone to be affected by human activities like urban development, industries, fisheries and tourism.

SCALES OF OCEANOGRAPHIC PROCESSES

In the process of studying marine biodiversity in a coastal system we need to be aware of the spatial and temporal dynamics of the physical environment.

Is the water temperature and the salinity we measured the day of the sampling responsible for the structure of the community we observed? This question is best related to the temporal variability of the physical conditions. Oceanographic processes could occur in a wide range of cycles, from (1) a short period, as the waves we see shoaling along the coast every three to four seconds and the sea level changes produced by tides every six to 12 hours, (2) medium period, like the seasonal changes in turbidity and productivity due to the effect of the seasons occurring in six month cycles, and (3) long period processes like the global circulation of a major current (such as the Gulf Stream), hurricanes and typhoons or El Niño events that occur every three to seven years. The shorter the period, the more evident the changes are to us.

We must also consider the spatial dynamics of the environment that surrounds our sampling site. The spatial variability of the physical variables on a shore could determine the structure of a community. Fresh water from a river might have more impact on intertidal communities if they are located in a place where the dominant littoral current transports the freshwater or the sediments. Exposed shores have different communities (in species composition and/or community structure) than protected ones.

KNOWING YOUR SITE, KNOWING YOUR SURROUNDINGS

It is always a good idea to have a birds-eye perspective of your sampling site. Go to the local cartography office and get a good map of your area. It is useful to have at least one map of a small scale ($\sim 1:100,000$) and other more detailed ($1:20,000$ or better). Recent aerial photographs are also valuable to identify features in the field. There are also free satellite images at high resolution (15 to 30 meters) at different band compositions available from the Internet or if you have more resources, you can acquire a very high-resolution (0.6–4m) image of your site from IKONOS or QuickBird satellites. Locate your site in the maps/photos/images, obtaining the GPS coordinates using the correct datum and projection for your maps. You can also map the surroundings communities or bottom types.

Next, identify all the oceanographic databases available for your region. Look for your local tide stations, meteorological stations, marine data stations, buoys, and gauge stations for local rivers (if applicable). Many oceanographic data exist from automatic and semi automatic remote sensors and most of them are low cost or freely available at the official local environmental offices or on the Internet (see list of further resources). However depending on the location of your site, it is probable that you will need to collect your own environmental data. If you plan to start an oceanographic time series on your NaGISA site, keep in mind that the oceanographic variables we measure during our sampling campaigns are the consequence of the combination of local and regional phenomena, in a temporal context. So, sea temperature or salinity taken once or twice in a year is probably inadequate to describe the environment of your site. If you have enough resources, small monitoring devices could be set in place. There are low price automatic meteorological stations and underwater temperature and light sensors. Always give thought to precision, accuracy and reliability of the instruments for your site, because the coastal zone is usually a very rough environment. A last consideration: if you plan to start a time series, plan also to maintain it for a long time.

MAIN OCEANOGRAPHIC VARIABLES IN THE NAGISA CONTEXT

Tides: Tides are the regular changes of the sea level produced by the gravitation pull of the moon and the sun on the water masses. The dynamics of the system are non-linear but well known since late 1700s. The harmonic constituents describe the interactions between the moon and the sun, considering many orbital constants and their effect on the water masses on the Earth. These coefficients can be estimated using the register from sea level gauge stations and they are regularly used to make tidal predictions. Tides also generate tidal currents, and in some embayment and semi-enclosed areas they can be the principle driving force of water circulation (Stewart 2004). Most countries have national offices that measure tides and produce tidal tables. Moreover, free software and Internet utilities for tidal prediction are available, but they must be used with extreme caution. It is highly advisable to use the official local tide data whenever possible.

Currents: In general, all movements of the water masses are referred to as marine currents. However, we can classify the movements considering their location and origin. In a local context, coastal morphology and the proximity of the sea floor are relevant, and they shape particular current systems. In a beach, littoral currents are the main force transporting the sediments. They originate with movement of the water and are defined by the shoaling of the breaking wave and the morphology of the beach. Littoral currents are the main constructors of the beach, continuously moving sediments in and out. Littoral currents are difficult to measure and they can change in direction and velocity in relatively short periods of time (weeks–months). However, the main pattern can be maintained for very long time. Coastal morphology gives important clues about the littoral currents, like the direction of the mouth of a small river or the sand pattern around a jetty.

Waves: Waves are undulations of the sea surface. Ocean waves have particular characteristics (wave-height, wave-length) that are modified when they reach the shallow coastal environment. The interaction with the sea floor causes the breaking of the wave and the ocean wave theory cannot be used in this situation. Normally, the waves we observe are caused by the wind stress over the sea surface, but other waves, often impressive in size and energy, are related to storm surges and earthquakes or submarine volcanic eruptions. The energy of the waves is a determinant factor in shaping the community structure and thus highly pertinent to the areas biodiversity. Different degrees of exposure and energy can be determined by observing wave patterns (Beer 1996).

Light: Light is of utmost importance to the great majority of primary producers. In rocky shores light is rarely a limiting factor, but it is for submerged vegetation communities. In water, light is scattered, absorbed and reflected. Depending on the wavelength, light attenuates rapidly with depth and after a few meters in coastal waters (5-30 m depending on suspended sediments and dissolved colored matter), less than 1% of the incoming light remains (Jerlov 1976; Bukata 1995). Several instruments can be used to measure the penetration of light (a photometer or radiometer for example), but the simplest one is the Secchi disk. Different Secchi disks give us different transparency values. It is important to always use the same disk and measure the transparency in the same conditions.

Ecosystem production: One fundamental variable of any community is its productivity. Primary productivity is the easiest and most common estimate and it is generally related to photosynthetic production. Although methods for estimating primary productivity exist, the concentration of photosynthetic pigments (such as chlorophyll) can be used as a proxy for the photosynthesis biomass produced by a community. The concentration of chlorophyll can change in response to changes in light and nutrients, as well as changes in phytoplanktonic community structure. Laboratory assays can determine pigment concentration, fluorometric *in situ* measurements, or remote sensing imagery. The last has the advantage of a high temporal resolution (one to two images per day) although it can be strongly affected by colored dissolved organic matter and suspended sediments.

Land inputs: Sediments from land can be a limiting factor for many communities, as they block the light and clog respiratory or alimentary mechanisms of marine organisms. Land run offs can also supply important nutrients to the sea. Suspended sediments can be observed using remote sensing imagery and its concentration can be estimated by simple laboratory determinations. Sediment traps can also be used and together with a carefully designed sampling plan, they can provide us with estimates of sedimentation rates. This variable is rarely found in any marine station data set.

Temperature: Temperature governs all metabolic processes. In general, animals and plants live within a relatively narrow temperature range. In the sea, extreme temperatures outside this normal range are rare, but seasonal variations can determine the presence of certain species over time. Measuring temperature is easy and several automatic sensors exist that can be deployed to obtain a continuous record. However, it is always necessary to calibrate sensors and take into account manufacturing calibration parameters in order to have a reliable data series. Daily sea surface temperature maps are available from the Internet derived from different satellite sensors (see list of further resources). Marine data stations routinely measure sea temperature and buoy data normally provide a depth profile.

Salinity: Salinity was originally defined as the content of salts dissolved in the water. In general and as in temperature, animals and plants live in a relatively narrow salinity range. In the open sea the salinity is usually around 36 parts per thousand (roughly 36g of salt in one liter of water), but in coastal waters, freshwater inputs can significantly lower this value. Nowadays salinity is defined operationally from its electrochemical measurement and it is expressed as practical salinity units (PSU). It can be measured with a conductivimeter or salinometer. Refractometers are also used although the results are less precise. Almost all marine data stations measure salinity.

FURTHER RESOURCES

- Edwards, A.J. 2000. Remote Sensing Handbook for Tropical Coastal Management. UNESCO Publishing
- Jerlov, N.G. 1976. Marine Optics. Elsevier Oceanography Series, 14. Elsevier.
- Open University. 1989. Ocean Circulation. Oxford: Pergamon Press.
- Open University. 1989. Seawater: Its Composition, Properties, and Behavior: Pergamon Press.
- Open University. 1989. The Ocean Basins: Their Structure and Evolution. Pergamon Press.

- Open University. 1989. Waves, Tides, and Shallow-water Processes. Pergamon Press.
- Stewart, R.H. Introduction to Physical Oceanography. Full text available at http://oceanworld.tamu.edu/ocean410/ocng410_text_book.html
- Tomczak, Matthias & J. Stuart Godfrey: Regional Oceanography: an Introduction. Full text available at <http://www.es.flinders.edu.au/~mattom/regoc/pdfversion.html>
- Thurman H.V. and A.P. Trujillo. Introductory Oceanography. 10th edition. Prentice Hall

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Section

20

Guide to Identification and Surveying of Nearshore Fishes

CHARACTERISTICS OF NEARSHORE FISHES

- 1) Associated with vegetation
- 2) Occur on sea bottom and in the water column
- 3) Vary in density and diversity with structural features of habitat

INTRODUCTION

Macroalgae and seagrass beds are dominant structural components in nearshore subtidal zones, providing habitat for many organisms. Assemblages of fishes are conspicuous in these vegetative habitats, using this biogenic structure for food and shelter. The most conspicuous macroalgae are kelps that form extensive canopies at the water surface (primarily giant kelp, *Macrocystis pyrifera*, and bull kelp, *Nereocystis leutkeana*) and seagrasses that occur very close to shore on open coastlines (surfgrass, *Phyllospadix* spp.) or in embayments (seagrasses, *Zostera marina* and other species). Several other brown macroalgae layer the bottom or form subsurface canopies 1–2m above the sea floor.

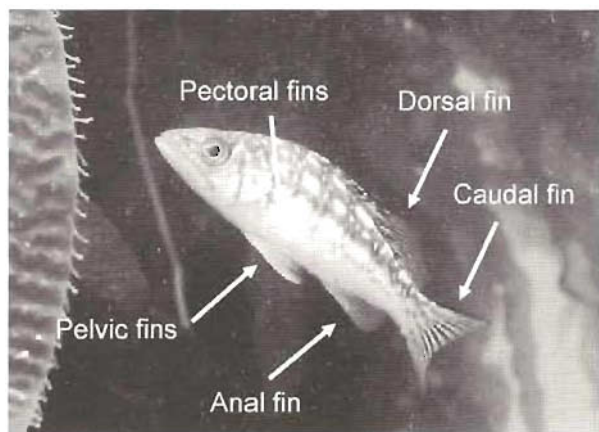
The fishes that reside in such habitats often vary in their density and diversity depending on the area, abundance, and structural features of vegetation. These fishes come in many different shapes, sizes, and with distinctive coloration that make their identification underwater possible.

GENERAL EXTERNAL FEATURES OF FISHES

Besides body shape and coloration, other identifying features relate to the color, size, and shape of fins. The pectoral and pelvic fins are *paired* fins, whereas the anal, dorsal, and caudal fins are *median* fins. The top, bottom, front, and back of a fish are the dorsal, ventral, anterior, and posterior regions, respectively.

CONDUCTING UNDERWATER TRANSECTS TO RECORD SPECIES AND DENSITY

Identification. Fishes occur in two main groups: **cartilaginous fishes** such as sharks, skates, and rays, and **bony fishes** (the typical fish). Within these



(photo courtesy of T. Anderson)

Fig. 20.1 Kelp Bass, *Paralabrax clathratus*

two main groups, fishes are generally organized and recognized according to family, for example: rockfishes (Scorpaenidae), basses (Serranidae), and surfperches (Embiotocidae), and within a family classified by genus and species. For underwater identification, many nearshore fishes can be distinguished by their shape, color, and pattern of coloration. Several excellent guides to the identification of fishes have been published (see Further Resources), and the use of keys for identification (for example, Miller and Lea 1972, Eschmeyer *et al.* 1983, Lamb and Edgell 1986, Love 1996) can be helpful if a fish can be examined closely or if preserved specimens are available. Familiarize yourself with the guides to the near shore fishes in your location.

Transects. To obtain a reasonable estimate of the density of fishes at a location, visual transects are most commonly used. Transects can be visualized as three-dimensional corridors through which fish pass. As a diver swims a transect, he/she should look ahead approximately 3–5m at a time and view the fishes within that corridor, taking a visual “snapshot” of the fishes present. Once the fishes in that section of the transect have been recorded, the diver does not look for fish again within that section; instead, the diver proceeds forward to take a “snapshot” of the next section of the transect, and so forth, until the transect is completed. It is important that divers swim a consistently slow-to-moderate pace (about 10 meters per minute) to record fish. Stopping to record data while swimming a transect is not advisable because fish are attracted to divers and a diver may record fish that moved into the transect corridor that were not there before or count the same fish more than once. Transects are used to obtain a density per unit area or volume, and “snapshots” along transects are used to avoid counting fish more than once as they may swim back and forth through a corridor.

To record the species and number of fish, each diver should carry an underwater slate with an attached pencil (underwater paper can be used, but data can be written directly on the slate). While the divers are recording species, they should also record the size of fish according to different bins. Such bins are designated size classes: <5cm, 5–10cm, 11–15cm, 16–20cm, 20–30cm, and >30cm. All divers performing transects should

first practice estimating the lengths of objects of various shapes and sizes. This is especially important to do underwater because objects are smaller than they appear! A very helpful guide is to glue a metric ruler to the slate or mark one edge of the slate in centimeters with a permanent marker.

Transect dimensions should be 2 meters wide by 2 meters high and 30 meters long. Four to six transects on the bottom and in mid-water should be done at 12 meters depth, and another four to six transects on the bottom and in mid-water should be done at 6 meters depth, totaling eight to 12 bottom and eight to 12 mid-water transects. These two depth contours should contain most if not all of the species that occur in macroalgae such as kelp beds. Of course, seagrass beds are generally shallower (<7–8 meters), so select depth contours that are relevant to fish surveys in this habitat (e.g. 2m and 6m). If the depth is the same within a kelp bed or seagrass bed, conduct the two sets of four to six transects at a reasonable distance from each other relative to the size of the bed. Seagrasses do not extend upward extensively into the water column, so conducting transects in the water column is not necessary.

For censusing macroalgal beds with algal canopies at the water surface, transects should be done along the bottom and in the water column by pairs of divers, with one diver recording fishes in the water column at 5–10m above another diver recording fishes along the bottom. Fishes are recorded for each transect (one through six) at each depth (6m or 12m). The diver on the bottom attaches a 30-meter fiberglass tape to the bottom using a clip or releasable cable tie attached to vegetation (algae or seagrass). After a transect has been completed along a depth contour (e.g. 12m), the bottom diver pulls on the transect tape to release the clip or cable tie from the vegetation and rolls up the tape on the reel while remaining stationary. The bottom diver then swims forward a short distance (about 5m) and again attaches the tape and follows the designated depth contour. The diver in the water column follows slightly ahead of the bottom diver while conducting a transect to avoid disturbing fishes in the water column that may move away from the rising bubbles produced by the bottom diver.

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Section 21 Sampling and Monitoring Rhodolith Beds

CHARACTERISTICS OF RHODOLITHS

- 1) Free living, non-geniculate coralline red algae
- 2) Globally distributed generally in sandy shallow water systems
- 3) Require water motion to remain in an unattached state

INTRODUCTION

Rhodoliths are free-living forms of non-geniculate coralline red algae (Corallinaceae, Rhodophyta) that form extensive beds worldwide over broad latitudinal and depth ranges (Foster 2001). Synonymous with the maerl beds common in the northeastern Atlantic, rhodolith beds are hard benthic substrates, albeit mobile, made up of branching crustose coralline thalli. Collectively they create a fragile biogenic matrix over carbonate sediment deposits thought to be the result of long-term accumulation of dead thalli (Minoura and Nakamori 1982; Steller and Foster 1995). To persist these algal beds require light, nutrients and movement from water motion (waves and currents) or bioturbation, which maintains them in an unattached and unburied state (Bosence 1976). A wide morphological variation of individuals exists and appears to be a response to variation in physical factors (Bosence 1976; Steller and Foster 1995). This variation in morphology and incorporation of whole rhodoliths and carbonates into the fossil record has led to their use as paleoindicators of environmental conditions (Foster *et al.* 1997). Despite their global representation in both living beds and fossil deposits, presently no world monograph exists for the taxonomy of rhodolith-forming species. However, the few existing accounts have both modern species descriptions and represent major geographic areas.

Rhodolith beds support a rich community of flora and fauna found to be higher in species diversity than soft-sediment benthos alone (Steller *et al.* 2003). Organisms within a bed can associate with the surface of algal thalli (epi-fauna/flora), within the branches (crypto-fauna/flora) or in the underlying sediments (in-fauna/flora) (Steller *et al.* 2003). Factors influencing diversity patterns include increased architectural complexity and grain size, reduced sedimentation (Grall and Glemarec 1997), seasonal variation (Ballesteros 1988) and reduced predation.

Unconsolidated rhodolith deposits have long been harvested for human use as soil amendment in European waters (Blunden *et al.* 1977). However,

recent studies have shown that beds are highly susceptible to anthropogenic disturbance such as trawling harvesting and reduced water quality (review in Birkett *et al.* 1998). Slow rhodolith growth (Rivera *et al.* 2003, Steller *et al.* 2003) combined with the negative impacts of burial makes recovery after disturbance predictably slow.

To increase the ecological understanding of rhodolith beds, they must be described and sampled in a rigorous, comparable way. A basic methodology for surveying rhodolith beds is introduced here. Methods are described to: 1) find and map rhodolith beds, 2) determine a sampling regime that provides accurate taxonomic, distributional and diversity estimates, 3) measure rhodolith growth rates, 4) set up long term monitoring to detect natural and/or anthropogenic changes.

SAMPLING METHODS

Locating and mapping a site: Subtidal rhodolith beds are commonly associated with both extensive areas of carbonate sediments (Steller and Foster 1995) and onshore deposits of fossilized rhodoliths (Foster *et al.* 1997). Locating rhodolith beds may be expedited by seeking local knowledge from fishermen, information from herbarium collections, and aerial observations. Locating a site can be enhanced using a benthic grab for exploration and SCUBA surveys for Subtidal exploration, verification and mapping. In all cases, have a buoy and a GPS to mark the location and enhance relocation. Remote mapping of rhodolith distribution at a site can be efficiently determined using sonar and mapping done using side scan sonar appears promising (Tsuji, 1993).

DEVELOPING AN OPTIMAL SAMPLING PROTOCOL: Planing is required to develop an optimal quantitative sampling protocol that minimizes the impact of sampling and combines methods to maximize sample design while maintaining unbiased sampling. The first priority is determining the population or community feature(s) to be described and then determining the number of samples needed to reasonably estimate a mean and variance. Described here is a combined method for an initial sampling of a variety of rhodolith bed features using depth stratified sampling at multiple sites within one bed to characterize population and species distributions within a bed. This example is for a typical bed that spans a depth gradient and ~1km of coastline. The design allows for unbiased characterization of the following features within the bed: rhodolith taxonomic distribution and makeup, rhodolith size frequency distribution and community composition. A description of how sample units are distributed in space will be followed by specifics for each feature.

SAMPLING EQUIPMENT NEEDED

- 30-m waterproof transect tapes
- Underwater slate with pencil that holds data sheets on underwater paper
- Quadrats for estimating density of mobile organisms (1m² collapsible) and rhodoliths (0.16m²)
- Random Point Quadrat (RPQ) bar for estimating percent cover of sessile organisms (1m pvc bar with a 1.5m rope with 10 knots tied at random distances)
- Pre-labeled collecting zip-loc bags
- GPS – for site relocation
- PVC stakes + sledge hammer for marking sites in soft-sediment

DISTRIBUTION OF SAMPLING UNITS IN SPACE: At three different randomly selected sites within the rhodolith bed, two 25 m transects are run parallel to shore, stratified by depth to characterize relationships between depth and rhodolith morphology and associated organisms. At four randomly assigned distances along each 25m transect, data or samples are taken to characterize the rhodolith species and size frequency and associated community. The 1m² quadrat is placed at each random distance and counts of all mobile macro-organisms (>1cm) are made within the quadrats. The RPQ bar is placed inside the quadrat and the data of the sessile organisms and substrate from under each knot is used to estimate the percent cover per quadrat. If rhodoliths are present, the 0.16m² quadrat is placed in a corner within two 1m² quadrats per transect and all live rhodoliths are carefully collected for size frequency analysis and taxonomic identification.

RHODOLITH TAXONOMY AND SIZE FREQUENCY: Collecting material for taxonomic purposes should be conducted in a method that allows for description of all possible morphological and reproductive variants. Therefore, extensive collections at each site should be made covering the range of growth-forms and reproductive phases. Collected material should be preserved immediately in formalin to best preserve taxonomically relevant structures. For optimal anatomical analysis reproductive plants with different morphologies should be embedded and sectioned. There are two general procedures, one with white resin (Woelkerling 1988) and other with paraffin (Riosmena-Rodriguez *et al.* 1999) that can be used equally and depend upon available facilities.

To identify the plants it is necessary to evaluate the morphology and anatomy, suggested regional resources for doing this are: temperate Northwestern Atlantic (Bird and McLachlan 1992), tropical Northwestern Atlantic (Littler and Littler 2000), Northeastern Atlantic and Mediterranean (Cabiocch *et al.* 1992; Irvine and Chamberlain 1994), Indian Ocean (Verheij 1993), Western Pacific (Woelkerling 1996a–e; Littler and Littler 2003) and Eastern Pacific (Riosmena-Rodriguez *et al.* 1999; Athanasiadis *et al.* 2004). These accounts should not be considered exhaustive for any area as new records and species are still often found. Critical monographic work is highly recommended as a parallel project to clearly understand the species boundaries. Voucher specimens should be taken from the randomly collected samples (method explained in previous section). At a minimum, take three to five individuals with conceptacles that cover all life stages and the range of variation in size and form.

An unbiased sampling design that samples the population to be characterized should be used to assess coralline species within a bed – not just haphazard collections. The samples should be dried in the shade, labeled with depth, location, date and collector information and wrapped individually. Information on the size frequency of the rhodolith population within the bed provides basic demographic information. For each rhodolith collection taken in the 0.16m² quadrats the number of thalli are counted in size classes of 1cm increments. Variation in branch density can be estimated from a subset of samples. Reproductive ‘effort’ can be estimated by measuring proportion of individuals with conceptacles.

SAMPLING OF RHODOLITH BED COMMUNITY: Many studies describing patterns of faunal associations in rhodolith beds have sampled rhodolith substrates using cores and suction dredges. These sampling methods mix

epibenthic organisms found only on the substrate surface with organisms found only inside the rhodoliths or sediments, thus losing information about substrate relationships of individual groups. A sampling regime that partitions organisms by sub-habitats provides a better understanding of the role of rhodoliths in the diverse community of associated organisms. Density and percent cover of common epibenthic macroflora and macrofauna are determined from the 1m² quadrats. Cryptofauna, the organisms between the branches of rhodoliths, can be determined from individual rhodoliths collected from the 0.16m² quadrats. Each rhodolith must be placed in a separate individual bag to retain associated organisms. Infaunal organisms found in sub-rhodolith sediments are sampled using small sediment cores taken within the 0.16m² quadrats, after living rhodoliths are removed. Both cryptofaunal rhodoliths and infaunal sediment cores should be preserved in 4% formalin solution for later sorting.

PHYSICAL MEASUREMENTS: Measures of physical parameters may be very important for understanding a rhodolith system. Features include: water clarity (use a secchi disk), water temperature (thermometer or temperature logger), salinity (salinometer), light (light meter), waves and currents (current meter).

ESTIMATING RHODOLITH GROWTH RATES: *In situ* coralline growth rates can be measured using the vital calcium carbonate stain, Alizarine Red (Rivera *et al.* 2003; Steller *et al.* 2003), which binds with surface epithelial layers down to about 4 cell layers. New apical growth occurs beyond this line and subsequent sectioning reveals the pink stain allowing new growth to be quantified. This method describes tagging, staining, recovery and growth rate measurements.

Collect replicate rhodoliths spanning the size range of interest and tag individuals prior to staining as branch tip breakage occurs during handling. Fragile species can be tagged with 1cm wide strips of stretchy pink flagging or surveyors with the date written on the strip. For durable species, plastic coated wire cable ties and fishing line with dated tags work well. Double tag all individuals and tag enough individuals to cover a 50% loss.

Mix Alizarin Red at a concentration of 0.25g/liter seawater and place tagged rhodoliths in the stain for 24 hours and aerate. Replace individuals, marking the subtidal location well. If samples will be left for an extended period of time or for areas of high current, rhodoliths may be put in 1×1×0.25m² PVC and plastic mesh corrals to enhance retrieval rates.

After the growth period, recover samples carefully to minimize the breakage of apical tips. Dry samples in shade, verify label and wrap individually in paper towels. Embed ten branch tips per plant in clear epoxy on a microscope slide, let dry, and then add another drop of epoxy. Grind down the surface with fine grinder to expose the stain and measure the new apical growth. Measure the distance from the stain to the tip to determine an average axial growth rate in mm/year. Double to give growth in diameter rate per plant in mm/year.

LONG-TERM MONITORING AND DETECTING CHANGE: The previously described sampling protocol should allow for detection of 'change' through time within a bed. An analysis of the initial data set can be used via power analysis to determine what future change will be detectable. To rigorously detect 'recovery' from a determined impact over time a BACI (Before-After-

Control-Impact) design should be followed (Stewart-Oaten 1986; Schroeter 1993). At minimum this includes one impact and one control site that are both measured before and after the impact. Numerous publications are available discussing proper sampling design (reviewed in Andrew and Mapstone 1983).

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Part VI

Reference Tools

Section 22

Priorities within the Protocols

The following are given as general guidelines, they are listed here to give you a sense of the priority the NaGISA project puts on different parts of the protocols.

SP = Scientific Personnel running a Core NaGISA Site

NP = Non Professional running a NaGISA Site

PRIORITY OF HEIGHT (ROCKY SHORES)

1. Intertidal zone (high, middle and low): Mandatory
2. Subtidal zone 1 (1m, 5m, 10m deep): Mandatory for SP, preferable for NP
3. Subtidal zone 2 (15m and 20m deep): Optional

PRIORITY OF QUADRATES

Rocky Shore Set

1. Temperature (surface/bottom), latitude and longitude: Mandatory
2. 1×1m – photograph and counts: Mandatory
3. 50×50cm – collecting: Mandatory for SP, preferable for NP
4. 25×25cm – collecting: Mandatory for SP, preferable for NP
5. Optional
 - Light, Salinity, pH
 - Include a 6th replicate for DNA analyses (preserve in 98% ethanol)
 - Plankton Pulls (see online protocols)
 - Fish Transects (Section 20)

Seagrass Set

1. Temperature (surface/bottom), latitude and longitude: Mandatory
2. 50×50cm (picture): Mandatory

3. 15cm core: Mandatory
4. 2cm core: Mandatory for SP, preferable for NP
5. Optional
 - 50×50cm: shoot counting in the field
 - Light, Salinity, pH
 - Include 6th replicate for DNA analyses (preserve in 98% ethanol)
 - Plankton Pulls (see online protocols)
 - Fish Transects (Section 20)

PRIORITY OF ANALYSIS

- Locality and physical information: Mandatory
- Species (presence) list: Mandatory
- Quantified data of selected taxa: Macroalgae/Seagrass (including dry and wet weights), mollusks, decapods, echinoderms, and corals: Mandatory
- Quantified data of other macrofauna: Mandatory for SP, preferable for NP
- Species list of meiofauna: Mandatory for SP, preferable for NP
- Quantified data of meiofauna: Mandatory for SP, preferable for NP
- Optional: Species list of plankton, fish and/or meiofauna from the 15cm core

DO NOT OMIT OR REDUCE

- Number of replicates – there should always be at least 5 replicates
- Photos – label with quadrat information, including latitude and longitude
- Do not choose unsuitable habitats as sampling sites, e.g., rocky shore without macroalgae, or soft bottom without seagrass
- Do not neglect to record taxa, record presence even if you can not identify

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Section 23 NaGISA Field Data Sheet

Documentation of a site's physical data, the percent coverage, number of species from the 1×1m quadrat and the recording of voucher specimen identity are extremely important. Once filled out, the example below would become the field data sheet for one replicate. It is prudent to keep the field data sheets even after imputing the data in to the summary data sheets (NaGISA Excel Data sheets, available from your regional coordinator).

NaGISA

ID number _____ Location _____
 Collection date _____ Time of collection (start) _____
 (Min) _____ (End) _____
 Collector(s) _____
 Type of habitat (circle one): macroalgae V. seagrass
 its Depth _____ Replicate number _____
 Height/Depth (cm) or ft in one: High Mid Low
 Temperature: surface (°C) _____ bottom (°C) _____
 Light: Surface _____ 1 _____ 2 _____ 3 _____ 4 _____ 5 _____ 6 _____ 7 _____ 8 _____ 9 _____ 10 _____ 11 _____ 12 _____ 13 _____ 14 _____ 15 _____ 16 _____ 17 _____ 18 _____ 19 _____ 20 _____

Voucher Species No./Species Name Sketch (to help you remember)	% Coverage or No. of Individuals	Voucher Species No./Species Name Sketch (to help you remember)	% Coverage or No. of Individuals

Section 24 Glossary of Methods

TECHNIQUES FOR PLANT MATERIAL

PRESSING (s): Seagrasses and algae are best stored as pressed specimens. If stored in good (dry) conditions, they can last several centuries. Although they are less suitable for anatomical studies than liquid-preserved specimens, the color and gross morphology are retained for much longer. DNA can be extracted from voucher specimens even after many years, if the specimens were not treated with formalin, and were quickly dried and stored properly. Place fresh, wet specimens on herbarium mounting paper and using as little water as possible spread the thallus out, displaying the characteristic branching pattern. Cover with gauze, blotter paper, non-stick wax paper or cloth, and press between absorbent material sheets; paper, newspapers or paper towels. Bind the sheets together in a herbarium press or simply stack one on top of another, bind with twine and weigh down with weights. Dry in a well-ventilated area at ambient room temperature. During the first week, due to the high water content of the specimens, the sheets of absorbent material have to be changed daily. The specimens should be dried for several weeks. Circulating air (fans, or simply allowing air to flow below, above and around the stack) will help dry the specimens faster. Well-dried specimens should be labeled with sampling information; species name, collector name, sampling place and date of collection. Wrap with a plastic sheet or put in a paper holder to protect from dirt.

FROZEN PRESERVATION: Place the specimen in a plastic bag, if possible laying it out flat. Specimens treated like this are suitable for DNA extractions, morphological observations and will not lose colour but consume considerable space and energy, and risk damage by mechanical failure of freezers or by power interruptions.

TECHNIQUES FOR BOTH PLANT AND ANIMAL MATERIAL

FORMALIN PRESERVATION: Formalin is a dangerous chemical, and should be handled in accordance with recommendations by health and safety agencies and the supplier; however it is a fast and effective way to preserve specimens. It is particularly useful in the field as solutions of less than 10% (1 part full-strength [= 39%] formaldehyde and 9 parts seawater) can be used, reducing the amount of liquid that needs to be carried in the field. Always use neutralized formalin in seawater and ensure that the specimens are not crowded in the jars to avoid the creation of formic acid, which can damage specimens. Specimens treated like this are suitable for later anatomical observations, but DNA extractions from formaldehyde-preserved specimens have not been very successful.

NEUTRALIZED OR BUFFERED FORMALIN: If the formalin solution is made up using seawater rather than fresh or tap water, the natural buffering capacity of the salts will help reduce the creation of acid which can lead to decalcification, discoloration and the eventual breakdown of specimens. However to ensure that the solution is not acidic it should be buffered. Buffer 1L of concentrated formalin by adding app. 15g CaCO_3 or Borax. Shake well (it will not completely dissolve).

ETHANOL PRESERVATION: Specimens can be placed directly in >75% ethanol. However, unless DNA analysis is going to be conducted (see *DNA preservation*) this is not recommended for soft tissue.

PRESERVING SAMPLES FOR MOLECULAR (DNA) ANALYSIS: If material is to be used for DNA analysis, it should be put directly in >80% ethanol. Pressed specimens and those preserved in Silica Gel (see preservation techniques) can also be used to save DNA for extraction and analyses. As water escaping from the sample will dilute the concentration of ethanol it is prudent to replace the ethanol within 24/48 hours.

SILICA GEL PRESERVATION: Small pieces (2g) chopped into less than 1mm of fresh dry algae, seagrass or sponge can be placed in an airtight container covered with silica crystals (electrophoretic grade). Specimens treated like this are suitable for DNA extractions although less so for morphological observations as they will shrink and change shape.

STORAGE: Specimens should generally be transferred to >70% ethanol for long-term storage. Ethanol (ethyl alcohol) is different from isopropyl alcohol, which is not recommended for long-term storage. A small amount of glycerol may be added to prevent specimens from drying out if there is a risk of the ethanol evaporating.

TECHNIQUES FOR ANIMAL MATERIAL

RELAXING /ANAESTHESIA: Relaxing specimens allows them to be worked with easier, reduce trauma, and is recommended when preparing taxonomic specimens in order to preserve subtle details and soft body forms. There are a number of ways to do this:

Magnesium salts: magnesium chloride MgCl_2 , magnesium sulfate MgSO_4 can be mixed with fresh or tap water in to which the specimen can then be placed, useful for polychaetes and echinoderms etc.

Chloral hydrate: add an isotonic solution of chloral hydrate to a container containing sufficient seawater for the animal to expand fully, useful for cnidarians

Menthol: an isotonic solution or crystals can be added to the seawater surrounding the specimens, useful for cnidarians and meiobenthos samples

Nicotine: crushed tobacco from a cigarette can be sprinkled on the surface of the water, useful for cnidarians.

Clove oil: add a few drops of clove oil in the seawater surrounding the specimen, useful for decapods and molluscs.

Propylene phenoxylol: add 1% *propylene phenoxylol* to the seawater surrounding the specimen, useful for decapods.

95% ethanol: can be slowly added to the seawater surrounding an animal placed in a small dish. After about an hour, a moderately sized echinoderm or mollusk will become sufficiently unresponsive that it can then be injected with fixative with little or no change in the orientation of soft parts such as tube feet.

Chilling: larger specimens, particularly from tropical regions, can be placed in seawater and left in the refrigerator overnight. The animal will die in a relaxed posture, and can then be fixed.

PRESERVATION BY DEHYDRATION: Effective for echinoderms (sea urchins, sea stars, and brittle stars), cnidarians (corals) and crustaceans (decapods). Specimen should be fixed in >70% ethanol for several days before drying in a well-ventilated area out of direct sunlight.

BLEACH DIGESTION (SPONGES): This technique is for rapid non-permanent preparations of spicules or sclerites. Place small fragments of sponge tissue (1–2mm) into a small tube, flask, or on top of a glass slide. Add a few drops of active bleach (sodium hypochlorite) and let the tissue dissolve completely. Wash once or twice carefully with water to eliminate the bleach before placing the slide on the microscope. If cleaner or permanent preparations are required it is more convenient but more tedious to use acid digestion.

ACID DIGESTION (SPONGES): This should be done in appropriate clothing and facilities able to deal with noxious acids (fume hood, protective glasses, container for used acid disposal, etc).

Place 0.5cm^3 of chopped sponge tissue into a test tube and cover with 2× volume of nitric acid, and either leave overnight in the hood or accelerate tissue digestion by placing the tube in a tube heater, or subjecting it to a slow flame such as an alcohol flame.

Once the solution is clear aspirate the acid with a pipette and, and proceed with a series of washes. Washes start with regular tap water and follow with a series of alcohol solutions (70%, 90%, 95%, 100% ethanol). For each wash the supernatant liquid has to be carefully aspirated after spicules settle at the bottom and discarded properly. After the last wash in 100% alcohol the mixture is gently swirled and a few drops placed onto a clean glass slide.

After the alcohol evaporates (a slide heater or a flame can be used to accelerate this process) spicules are mounted permanently with an appropriate mounting medium (e.g., Canada Balsam, Permount, Depex, Euparal etc.).

SECTIONING (WET SPONGE): Using a (wet) specimen that includes surface and core sponge tissue and has been preserved in 1–2 cc alcohol thinly (0.2–0.5mm) cut sections from the surface to the core of the sponge by hand using a new razor blade. Dehydrate the sections through a series of 95% ethanol (four times, 3–5 min each), and 100% ethanol (twice, 3–5 min each). Place the sections on a glass slide. Add a drop of toluene (or other clearing agent, carefully as toluene is toxic) so that it just covers the section. Add a few drops of Permount or Canada Balsam. Dip one side of a cover slide into toluene (decreases the formation of bubbles) and place it gently on top of the sections embedded in the medium. If the sections are too thick a piece of monofilament (fishing line) on each side of the sections can help to place the coverslip. The mounting medium takes 24–72 hours to dry. Placing it on a heating plate (medium to low heat) accelerates the drying of the medium.

SECTIONING (DRY SPONGE): A piece of a specimen can be air-dried overnight or in an oven (60–65°C) for a few hours. Sections made on dried pieces are easier to cut, and do not need to go through the dehydration steps given above; dry sections can be placed directly in alcohol (100%), then moved to toluene and directly mounted. Several skeletal arrangements can be studied in this way.

Section 25 Glossary of Terms

Abdomen. In crustaceans this is the posterior part of the body (the second region of the two part body plan of decapods and the third region of the three part body plan of amphipods) consisting of six segments, sometimes including a tail fan. In shrimps and lobsters the *abdomen* is well extended, thick and strong. In crab-like species, the *abdomen* is flattened, reduced in size and fold under the body.

Ambulacrum. The region that runs along the oral surface of an echinoderm's *ray* containing the *tube feet*.

Annuli. Segments of the *flagellum* or *antennae* (often erroneously called articles or segments).

Antenna(e) (1 & 2). Appendages found on the *head*. In shrimps and lobsters they are long, wipe-like, sometimes spiny; in crabs these are shorter, sometimes inconspicuous but in all crustaceans, they are the first (antennule) and second (antenna) appendages and are comprised of a *peduncle* and *flagellum* of various length; and may have antennal scales and *annuli*. In polychaetes, antenna refers to the sensory appendages arising from the anterior end or dorsal surface of the *prostomium*.

Appendage(s). A 'limb-like' extension from the main body, including legs, *rays*, *antennae* and antennules.

Appendix masculina. A stylet-like copulatory structure on the inner margin of the *endopod* of a male crustaceans' second *pleopod*.

Area. A distinct geographic region. The NaGISA plan outlines that 3 *areas* should be chosen so that they combine to represent the maximum biodiversity found along the shoreline within each project specified 20° box (20° latitude × 20° longitude). Each area is to have at least 3 *sites*.

Article. Segment of an arthropod limb, usually articulating.

Ascocyst. A specialized vegetative cell in brown algae (e.g. Scytosiphonales), occasionally called a paraphysis.

Axial. In gastropods, this refers to the line more or less parallel to the axis of coiling.

Beak. The small tip of a bivalve shell, near the *hinge*.

Benthic. Refers to all marine species that live on the bottom (sea floor), either in shallow water (intertidally or subtidally: on beaches or rocks, in coastal lagoons, among seagrasses, in coral reefs) or in deep water. These species belong to the benthos and are divided in two major groups: species that live above the substrate and are part of the epifauna and those that live burrowed in the sediments and are part of the infauna.

Body whorl. The last *whorl* of a snail shell.

Branchia. Gills – respiratory organs that remove dissolved oxygen from the surrounding water. In arthropods they are located symmetrically on both sides of the *cephalothorax* or *thorax*.

Branchial cavity/chamber. Space between thorax and lateral flap of the *carapace* enclosing *branchia* or gills in some crustaceans e.g. decapods.

Callus. A thick deposit of smooth shell material or enamel in mollusks.

Calyx. The central cup that contains the gut and from which the feeding *appendages* radiate in crinoids and some stalked, fossil echinoderms.

Cancellate. Mollusk shell sculpture consisting of crossing lines.

Carapace. A shield consisting of hard (strongly calcified) or rather soft (poorly calcified) tegument covering the *head* and the thoracic *appendages* of crustaceans.

Carpospore. Diploid reproductive cell of red algae formed by the *carposporophyte* on the female gametophyte.

Carposporophyte. A generation of red algae developed on the female gametophyte after fertilization; forms a *carpospore*.

Cephalothorax. The fused segments of the head and thorax, covered by the carapace in decapods.

Chaetae. A chitinous rod-shape process arising from the *notopodia* and/or *neuropodia* of the *parapods* of polychaetes.

Chela. Claw or pincer, located at the end of a leg in crustaceans comprised of a fixed finger and a moveable finger.

Choanocyte. Flagellated cell type found in sponges, with a characteristic set of cytoplasmatic projections that form a net-like collar, essential to filter very small (<2µm) dissolved organic matter from the water. The beating flagella pump water through the sponge at rates of up to 1 l/sponge cc/hour.

Coenocyte. Large multinucleate cell constituting the entire *thallus* of an individual alga.

Columella. The central column around which the gastropod's shell coils.

Concentric. In bivalves this refers to the direction coinciding with that of growth lines.

Continental shelf. Sea floor extending between the low water line and (usually) about 150–200 m depth, where most fishing activities occur.

Core/Corer. A cylindrical tube (corer) of defined radius (15cm or 2cm) that is wedged into the soft sediment to a defined depth (10cm) and removed with the sediment intact (core), the sediments is then sifted and sorted for specimens.

Depth contour (Transect). An imaginary line connecting the ground of the same height/depth. A *transect* following the depth contour follows the undulations of the shoreline and thus is very rarely a 'straight' line.

Endopod. Inner *ramus* (branch) of a limb, paired with the *exopod*, in crustaceans.

Es. An index of biodiversity. Usually followed by two numbers as in Es (50) = 25, the number in brackets refers to the number of randomly selected specimens from the area in question and the second number refers to the number of unique species expected to be found amongst those specimens.

Exopod. Outer *ramus* (branch) of a limb paired with the *endopod*, in crustaceans.

Flagellum. Distal part of a crustaceans *antenna* 1 or 2, generally made of many *annuli*.

Gamete. Sexual reproductive cell.

Gametophyte. A haploid generation that forms male and/or female *gametes* in algae.

Gland cell. A specialized vegetative cell in red algae, involved in secretion or storage.

Gnathopod. First or second *pereopod* in amphipods, differing in function and appearance to the following *pereopods*, usually *chelate* or *subchelate*.

Habitus. General appearance, body build and constitution of an organism.

Head. Anterior section of the body. In crustaceans it is the first of two or three regions of body, incorporating segments bearing pairs of antennae and mouthparts.

Hinge. In bivalves, the interlocking, often toothed, structure in the dorsal region of the valves.

Hydropore(s). Minute pores, usually contained within the *madreporite*, that lead to a canal that connects internally to the *water ring* of the *water vascular system* of echinoderms. Hydropores are believed to replenish water lost through the *tube feet*.

Incisor process. Distal biting part of a crustaceans mandible, typically found with teeth.

Lacinia mobilis. Enlarged, nearly articulated spine of a crustacean's mandible *spine row*, adjacent and similar to the *incisor process*, found on the left side of the *mandible*.

Lamella. A thin plate or ridge often appearing in multiples.

Lip. In gastropods, the margin of the aperture.

Madreporite. A 'sieve-plate', finely pierced by *hydropores*, embedded in the body wall of sea stars, sea urchins, brittlestars, and held within the coelom of sea cucumbers. The *madreporite* is connected to the *water ring* by the *stone canal*.

Manca. One of the first three stages of a Peracarida's (e.g. isopod) postmarsupial life cycle, recognized by having the seventh *pereopod* absent or rudimentary.

Mandible. Third limb of a crustacean's *head* (after *antennae*), and first mouthpart; important components are the *incisor process*, *spine row*, *molar process* and (occasionally) articulated palps on the left and right sides, and *lacinia mobilis* on the left side.

Marsupium. A ventral *pereonal* brood-pouch on female amphipods and isopods for developing embryos composed of oostegites projecting from the coxae of anterior *pereopods* and sometimes from the *maxilliped*.

Maxilla (1 & 2). 1- The fourth (maxillule) limb or second mouthpart on the *head* of crustaceans, comprising of a large outer lobe (endite) armed with short tooth-like setae, and a smaller inner lobe (endite) with 1–3 longer setae. 2- The fifth (maxilla) limb or third mouthpart on the head of crustaceans, comprising of a basal segment usually bearing three setose lobes (endites).

Maxilliped. Counted as the sixth limb on the *head* it is actually the first limb of a crustacean's thorax. It is the fourth mouthpart, comprising of the coxa, the basis bears an extended setose endite *palp* with 5 articles, and an epipod attached laterally to the coxa and fused to the head.

Medusa. Solitary, typically pelagic stage of the cnidarian life cycle.

Meristem. A cell or group of cells that divide conspicuously and more actively than other cells (regions), with an essential role in constructing the *thallus* in alga.

Metamorphosis. Transformation, usually radical and with extensive remodeling and resorption of tissues, of the newly settled larval stage into a miniature version of the adult.

Molar process. Grinding portion on the inside of a crustacean's *mandible* (usually armed with ridges, teeth and *setae* although these are sometimes reduced or absent).

Mutable collagenous tissue. Connective tissues capable of changing stiffness in response to nervous control. Found in the body wall of echinoderms.

Nematocyst. Intracellular stinging capsule used by cnidarians in offense and defense; the *sine qua non* of cnidarians.

Neuropodia/Neuropodium. Ventral *ramus* of a *parapodium* in polychaetes.

Nodulose. The adjective used to describe a structure with small knot/knob like protuberances or outgrowths.

Notopodia/Notopodium. Dorsal *ramus* of a *parapodium* in polychaetes.

Operculum. A gastropod's 'trap door' which closes off the shell from the external environment when the animal retracts.

Oscule(a). Ex-current aperture of the filtering system of sponges. Usually visible, ranging in width from 1mm to several cm. Its size, position and shape are important morphological characteristics.

Palp. In crustaceans this is the lateral *appendage* of the *mandible* (maximum 3 articles) or maxilliped (maximum 5 articles). In polychaetes the palp is a paired appendage emerging from the *prostomium* or *peristomium*, with a sensory or feeding function.

Parapodia/Parapodium. Segmented foot-like projection of polychaetes, usually bearing *chaetae*.

Parietal wall. The area on a gastropod's *whorl* near the *columella*, opposite the outer *lip*.

Pedicellariae. Minute, jawed structures made of *stereom* found on the external surface of echinoids and asteroids.

Peduncle (protopod or sympod). Basal segment of a crustaceans *pleopods* and *uropods* from which the flagellum or *endopod* and *exopod* arise.

Pelagic. Refers to all marine species that are free-swimming in the mid-water water mass, by definition pelagic species are not associated with the sea floor.

Pentamery. The basic 5-part, radial symmetry typical of adult echinoderms as manifested by the *rays* that extend outward from the mouth.

Percent cover. The degree of which a species covers an *area*, in the NaGISA case it most often refers to the relative amount of the species found in the 1×1m quadrat in a replicate set. Complete coverage equals 100%, and care should be made to separate the overhead or canopy coverage from the *benthic* coverage (they are given two separate columns in the field data file) and to record the percentage of bare rock as well.

Pereon. Second of three regions of the body of amphipods and isopods.

Pereonite. One of seven segments of the isopod and amphipod *pereon*, each bearing a pair of *pereopods*.

Pereopod(s). A limb of the *pereon* or *cephalothroax*, comprising of 7 articles (or segments), named from the proximal end: coxa, basis, ischium, merus, carpus, propodus, dactylus. In amphipods and isopods *pereopods* are numbered 1 to 7 and are equivalent to limbs 2 to 8 of the thorax. In decapods there are five pairs, the first pair ends in a claw, in some groups, legs 4–5 or just 5 are reduced in size.

Periostracum. The outermost layer of the molluscan shell composed of a horny organic substance.

Peristomium. The segment behind a polychaete's *prostomium*.

Periphery (of a whorl). The edge of a gastropods *whorl*.

Phaeophyceae. Brown algae. A member of Ochrophyta (Chromista) with multicellular thalli, using chlorophylls *a*, *c* and fucoxanthin (the source of its characteristic brown color) as its major photosynthetic pigments. Mostly marine. About 2,000 species in 300 genera are described worldwide.

Pleon. Third of three regions of the body of isopods. Second of two regions of the body of decapods.

Pleonite. One of first of (usually) 5 segments of the *pleon* or *abdomen* of crustaceans. See *pleotelson*.

Pleopod(s). One of the first (usually) 5 paired, biramous limbs of the *pleon* or *abdomen* of crustaceans.

Pleotelson. Segment derived by fusion of sixth *pleonite* and telson in some crustaceans e.g. isopods and tanaids.

Plication. A fold in a molluscan shell.

Polyp. Typically *benthic* stage of the cnidarian life cycle; may be solitary or colonial.

Proboscis. The anterior part of a polychaete's prestomodeum (gut), which can (usually) be everted past the *prostomium* for feeding and/or burrowing.

Protoconch. The first, usually uncalcified, translucent, smooth *whorls* of a gastropod's shell, forming the apex.

Pygidium. The post segmental, most posterior section of polychaetes, surrounding the anus.

Prostomium. The pre-segmental, most anterior lobe of polychaetes.

Quadrat(s). A square of a determined size (1×1m, 50×50cm, 25×25cm), which is used to define the ground to be sampled. Often made out of simple PCV tubing connected together and weighted for underwater use. Sections within the quadrat can be measured off with string to assist with calculating the *percent coverage*.

Quadrat set. A group of *quadrats* (one of each size: 1×1m, 50×50cm, 25×25cm) in rocky shores, or *quadrats* and *cores* (one of each size 50×50cm, 15×10cm, 2×10cm) in seagrass beds, that make up one of five of the replicates to be taken along a single *depth contour transect* during NaGISA field sampling.

Radial. In bivalves, in the direction of growth outward from the beak.

Radial canal. The part of the *water vascular system* of echinoderms that progresses from the circum-esophageal *water ring* along the *ray*, giving rise to the *tube feet*.

Ramus. A branch of a limb. In crustaceans these are the *endopod* and *exopod*, in polychaetes these are the *neuropodia* and *notopodia*.

Ray. That part of an echinoderm's body that radiates from the mouth and is accompanied by a *radial canal* and its *tube feet*. See *water vascular system* and *ambulacrum*.

Reticulate. A network of crossing lines in molluscan shells.

Rhodophyceae. Red algae. Traditionally divided into two subclasses, Bangiophycidae having simple *thallus* structures and life histories, and Florideophycidae having more elaborate *thallus* and apparent three-phase life histories. Chlorophyll *a* and phycobilin proteins are the major photosynthetic pigments. About 5,500 species in 600 genera are described worldwide, most of which are marine.

Rostrum. Prolongation of median part of anterior carapace between a crustacean's eyes. Can be inconspicuous, long or short, spiny or smooth.

Rudiment. A collective term for the structures formed in the earliest stages of a developing echinoderm larva. The rudiment becomes more strongly expressed as it interacts with various body cavities and tissues to develop a miniature version of the adult body form on the left side of the larva.

Seta. A cuticular process clearly articulated with the basal cuticle; often complex, with secondary setules or comb-like structures. Some literature uses the term 'spine' for thickened setae but 'robust seta' is more accurate. For polychaetes see *chaetae*.

Site. A distinct local ranging from 10 to 75 m hosting a continuous habitat. NaGISA *sites* within an *area* must be separate and distinct from each other. *Sites* are chosen according to the following qualities: Pristineness, Continuousness, Stability, Accessibility, and Relation to historic sampling areas or communities.

Slate. A rectangular board made of PVC, often sanded so that it can be written on with a pencil. Some slates are made to hold underwater paper to record data.

Spicule(s). Mineral structures secreted by many sponge species composed of either calcium carbonate or silica. Silicate *spicules* are secreted over a proteinaceous filament. Sizes range from a few micrometers to a meter. Its main function is to give strength, and support to the sponge body.

Spine row. Row of spines on mesial side of a crustacean's mandibular *incisor process*, proximal to *lacinia mobilis*.

Spiral. Following the same direction as the turning of a gastropod's *whorls*.

Spire. The visible part of all the *whorls* of the gastropod's shell except the last or *body whorl*.

Sporophyte. A diploid generation that forms asexual reproductive cells in algae; when meiosis occurs in the formation of reproductive cells, the cells will develop as haploid *gametophytes*.

Statocyst. Sac-like balance organ, usually containing a granule(s). It is found in the telson of anthuroids, the *antennae* of mysids and just behind the eye of many cephalopods.

Stereom. Usually porous, sponge-like calcium carbonate (limestone, or calcite) elements found in the body of an echinoderm. Although any given *stereom* plate will act optically as if it were a single calcite crystal, *stereom* is in fact polycrystalline, and also incorporates a proteinaceous matrix.

Stone canal. A tube, often invested with *stereom* elements, connecting the *madreporite* to the *water ring* of the *water vascular system* of echinoderms.

Subchelate. Claw-like extremity of a crustacean leg, forming an imperfect pincer with a moving finger closing in a vertical plane.

Taxon (pl. taxa). General term for a taxonomic entity of any taxonomic rank e.g. form, species, genus, class.

Tetraspore. Asexual reproductive cell principally formed after meiosis, four in each tetrasporangium. Widely seen in red algae and one order of brown algae i.e. Dictyotales.

Thallus. General term for a plant body without obvious differentiation i.e. the part not divided into blades/leaves, holdfasts/roots etc. It is often thin and flattened, as in the body of alga or the simple plant body of some seagrasses.

Transect. A (usually) straight-line corridor of varying dimensions (width, height, and length) used to record the density of organisms. When run subtidally it is referred to as an underwater *transect*. NaGISA transects follow the *depth contour* line.

Tube foot (feet or podia). The hollow, tube-like external manifestations of the echinoderms *water vascular system*, operated by exploiting the properties of a hydrostatic skeleton. Muscles extend the *tube foot* by squeezing water into the internal cavity to increase volume by increasing length. Opposing muscles for retraction operate to pull the tube foot back towards the body.

Ulvophyceae. A member of Chlorophyta (green algae). Chlorophyll *a* and *b*, and xanthophylls are the major photosynthetic pigments. About 1,100 species divided amongst 100 genera are described worldwide.

Umbilicus. The hole in the center of the basis of some gastropod shells.

Umbo. The upper or early part of the bivalve shell, opposite the *hinge*.

Unguis. Claw-like *seta* on the tip of dactylus of crustaceans.

Uropod. Most posterior paired limb of a crustacean, it is attached to the pleotelson in isopods with which it can form a tail-fan or be found in the styliiform or operculiform.

Varix. A thickened rib on the surface of a gastropod's shell marking the end of a growth stage.

Water ring. The part of an echinoderm's *water vascular system* that forms a ring around the esophagus, and which gives rise to the *radial canals*.

Water vascular system. A series of tubes in a echinoderm consisting of a circum-esophageal *water ring* that gives rise to the *radial canals*. Each *radial canal* runs the length of a *ray*, and supplies the external manifestation of the water vascular system, the *tube feet*. The water vascular system arises from the hydrocoel in the *rudiment*.

Whorl. A complete turn of a gastropod's shell.

Zoospore. A motile asexual reproductive cell produced by algae.

Section 26 Bibliography

Abbott, A.A., G.J. Hollenberg (1976) *Marine Algae of California*. Stanford University Press, Stanford, CA.

Abbot, R.T. (1974) *American Seashells*. Van Nostrand Reinhold New York.

Abbott, R.T. (1989) *Shells*. Portland House New York.

Abbot, R.T. and P. Dance (1982) *Compendium of Seashells*. Dutton New York.

Abbot, R.T. and P.A. Morris (1995) *Peterson Shells of the Atlantic & Gulf Coasts & The West Indies*. Field Guides. Houghton Mifflin Co. New York.

Aladro-Lubel, M.A., M.E. Martínez-Murillo, I.E. Lira-Galera and V.E. Rojas-Ruíz (1992) *Guía de Practicas de Campo: Protozoarios e Invertebrados de Estuarios y Marinos*. AGT, México, 101pp.

Anderson, M.J. (2001) A new method for non-parametric multivariate analysis of variance. *Austral Ecology* 26: 32–46.

Anderson, M.J., C.E. Diebel, W.M. Blom and T.J. Landers *In press* Consistency and variation in kelp holdfast assemblages: spatial patterns of biodiversity for the major phyla at different taxonomic resolutions. *Journal of Experimental Marine Biology and Ecology*.

Athanasiadis, A., P.A. Lebednik and W.H. Adey (2004) The genus *Mesophyllum* (Melobesioideae, Corallinales, Rhodophyta) on the northern Pacific coast of North America. *Phycologia* 43(2): 126–165.

Brodie, J.A., and L.M. Irvine (2003) *Seaweeds of the British Isles I. Rhodophyta; II. Chlorophyta; III. Fucophyceae (Phaeophyceae)*. The Natural History Museum and Intercept Ltd.

Avise, J.C. (2004) *Molecular markers, natural history and evolution*. 2nd Edition. Sinauer, Sunderland, 350pp.

- Ballesteros, E. (1988) Composicion y estructura de los fondos de *maerl* de Tossa de Mar (Gerona, Espana). *Collect. Bot. (Barcelona)* 17(2): 161–182.
- Barnard, J.L. and G. Karaman (1991) The Families and Genera of Marine Gammaridean Amphipoda (Except Marine Gammaroidea). *Records of the Australian Museum*, Supplement 13 two parts: 1–866.
- Beer, T. (1996) *Environmental Oceanography*. CRC Press.
- Beesley, P.L., G.J. Ross and C.J. Glasby Eds. (2000) *Polychaetes & Allies: The Southern Synthesis. Fauna of Australia. vol. 4A Polychaeta, Myzostomida, Pogonophora, Echiura, Sipuncula*. CSIRO Publishing: Melbourne, Australia.
- Beesley, P.L., G.J. Ross and A. Wells Eds. (1998) *Fauna of Australia vol. 5: Mollusca: The Southern Synthesis*. CSIRO Publishing Melbourne, Australia.
- Benedetti-Cecchi, L. (2001) Variability in abundance of algae and invertebrates at different spatial scales on rocky sea shores. *Mar. Ecol. Prog. Ser.* 215: 79–92.
- Benedetti-Cecchi, L., L. Airoidi, M. Abbiati and F. Cinelli (1996) Estimating the abundance of benthic invertebrates: a comparison of procedures and variability between observers. *Marine Ecology Progress Series* 138: 93–101.
- Bergquist, P. (1978) *Sponges*. University of California Press. Gran Bretaña, 268pp.
- Bird, C.J. and J.L. McLachlan (1992) *Seaweed Flora of the Maritimes 1 Rhodophyta — Red Algae*. Biopress Bristol, 177pp.
- Birkett, D. and C. Maggs and M. Dring (1998) *Maerl (vol. V): An overview of dynamic and sensitivity characteristics for conservation management of marine SACs*. Scottish Association for Marine Science (UK Marine SACs Project), 116pp.
- Blunden, G., W. Farnham, N. Jephson, R.H. Fenn and B.A. Plunkett (1977) The composition of *maerl* from the Glenan Islands of Southern Brittany. *Botanica Marina* 20: 121–125.
- Borchiellini, C., C. Chombard, M. Manuel, E. Alivon, J. Vacelet and N. Boury-Esnault (2004) Molecular phylogeny of Demospongiae: implications for classification and scenarios of character evolution. *Mol. Phylogenet. Evol.* 32(3): 823–837.
- Bosence, D.W.J. (1976) Ecological studies on two unattached coralline algae from western Ireland. *Palaeontology* 19(2): 365–395.
- Botosaneanu, L. Ed. (1986) *Stygofauna Mundi: A faunistic, distributional, and ecological Synthesis of the world fauna inhabiting subterranean waters (including the marine interstitial)*. E.J. Brill/Backhuys, Leiden. The Netherlands, 740pp.
- Boury-Esnault, N. and K. Rützler (1997) *Thesaurus of sponge morphology. Smithsonian contributions to Zoology* 596: 1–55.

- Bousfield, E.L. (2001) An updated commentary on the phyletic classification of the amphipod Crustacea and its applicability to the North American fauna. *Amphipacifica* 3(1): 49–119.
- Bridson, D. and L. Forman (1992) *The Herbarium Handbook*. Revised edition. Royal Botanical Gardens Kew, 303pp.
- Bruce, N.L. (1986) Cirolanidae (Crustacea: Isopoda) of Australia. *Records of the Australian Museum, Supplement* 6: 1–239.
- Brusca, G.J., and R.C. Brusca (2003) *Invertebrates*. 2nd Edition. Sinauer Ass. Inc. Sunderland, Ma., 936pp.
- Brusca, R.C. (1980) *Common intertidal invertebrates of the Gulf of California*. 2nd Edition. University of Arizona Press, Tucson 312pp.
- Brusca, R.C. (1983) A monograph on the isopod family Aegidae in the tropical eastern Pacific. I. The genus *Aega*. *Allan Hancock Monographs in Marine Biology* 12: 1–39.
- Bukata, R.P. (1995) *Optical Properties and Remote Sensing of Inland and Coastal Waters*. CRC Press.
- Cabioch, J. (1992) *Guía de las Algas de los Mares de Europa: Atlantico y Mediterraneo*. Omega, España 249pp.
- Chapin III, F.S. and O.E. Sala (1998) Ecosystem consequences of changing biodiversity. *BioScience* 48: 45–52.
- Clarke, K.R. (1993) Non-parametric multivariate analyses of changes in community structure. *Austral Ecology* 18: 117–143.
- Coles, R.G., W.J. Lee Long and R.A. Watson (1993) Distribution of seagrasses, and their fish and Penaeid prawn communities, in Cairns Harbour, a tropical estuary, Northern Queensland, Australia. *Aust. J. Freshwater Res.* 44: 193–210.
- Colwell, R.K., C.X. Mao and J. Chang (2004) Interpolating, extrapolating, and comparing incidence-based species accumulation curves. *Ecology* 85: 2717–2727.
- Cook, C.D. (1996) *Aquatic Plant Book*. SPB Academic Publishing, Amsterdam, 228pp.
- Cookson, L.J. (1991) Australasian species of Limnoriidae (Crustacea: Isopoda). *Memoirs of the Museum of Victoria* 52: 137–262.
- Dales, R.P. (1962) The polychaete stomodeum and the interrelationship of the families of the Polychaeta. *Proceedings of the Zoological Society of London* 139: 289–328.
- Dayton, P.K. (1975) Experimental studies of algal canopy interactions in a sea otter-dominated kelp community at Amchitka Island, Alaska. *Fishery Bulletin* 73: 230–237.
- de Vos, L., K. Rützler, N. Boury-Esnault, C. Donadey and J. Vacelet (1991) *Atlas of sponge morphology*. Smithsonian Institution Press: Washington and London: i–xi, 1–117.

- Dearn, S.L. (1987) The fauna of subtidal articulated coralline mats: competition, dynamics, and effects of spatial heterogeneity. MS Thesis. California State University Stanislaus, 51pp.
- den Hartog, C. (1970) The Sea-Grasses of the World. North-Holland Publ. Co., Amsterdam, 275pp.
- Dethier, M.N. (1984) Disturbance and recovery in intertidal pools: maintenance of mosaic patterns. *Ecological Monographs* 54: 99–118.
- Dethier, M.N., E.S. Graham, S. Cohen and L.M. Tear (1993) Visual versus random-point percent cover estimations: 'objective' is not always better. *Marine Ecology Progress Series* 110: 9–18.
- Díaz, Juan M. and Mónica Puyana (1994) Moluscos del Caribe Colombiano, Invenmar-Fundación Natura-Colciencias Bogotá, Colombia.
- Edwards, M. (2004) Estimating scale-dependency in disturbance impacts: El Niños and giant kelp forests in the northeast Pacific. *Oecologia* 138: 436–447.
- Eldredge, L.G. and C.M. Smith (2001) Guidebook of Introduced Marine Species of Hawaii. *Bishop Museum Technical Report* 21: 1–70.
- Erftemeijer, P.L., A. Djunarlin and W. Moka (1993) Stomach content analysis of a dugong (*Dugong dugon*) from South Sulawesi, Indonesia. *Aust. J. Freshwater Res.* 44: 229–233.
- Eschmeyer, W., E. Herald and H. Hammann (1983) A field guide to Pacific coast fishes (a Peterson field guide). Houghton, Mifflin, New York, N.Y., 336pp.
- Estes, J.A., M.T. Tinker, T.M. Williams and D.F. Doak (1998) Killer whale predation on sea otters linking oceanic and nearshore ecosystems. *Science* 282: 473–476.
- Fauchald, K. (1977) The Polychaete Worms: Definitions and keys to the orders, families, and genera. *Natural History Museum of Los Angeles County Science Series* 28: 1–188.
- Fortes, M. (1995) Seagrasses of East Asia: Environmental and Management Perspectives. ECU/EAS Technical Report Series No. 6. United Nations Environmental Programme, Bangkok, Thailand 79pp.
- Fortes, M.D. (1990) Taxonomy and distribution of seagrasses in the ASEAN Region. In M.D. Fortes and Nuning Wirjoatmodjo Study No. 6 Seagrass Resources in Southeast Asia, UNESCO — Jakarta (ROSTSEA) 15–57.
- Foster, M.S., C. Harrold and D.D. Hardin (1991) Point vs. photo quadrat estimates of the cover of sessile marine organisms. *Journal of Experimental Marine Biology and Ecology* 146: 193–203.
- Foster, M.S. (2001) Rhodoliths: between rocks and soft places. *Journal of Phycology* 37: 659–667.
- Foster, M.S., R. Riosmena-Rodriguez, D.L. Steller and W.J. Woelkerling (1997) Living rhodolith beds in the Gulf of California and their implications

for paleoenvironmental interpretation. In Pliocene carbonates and related facies flanking the Gulf of California, Baja California, Mexico *Eds.* M.E. Johnson, J. Ledesma-Vazquez. *Geological Society of America Special Paper* 318: 127–139.

Fritsch, F.E. (1945) The Structure and Reproduction of the Algae. Vol. I, II. Cambridge University Press, Cambridge.

Gaston, K.J. (1996) What is biodiversity? In *Biodiversity: a biology of numbers and difference* *Ed.* K.J. Gaston. Blackwell Science Ltd. Oxford p. 1–9.

Giere, O. (1993) Meiobenthology: The microscopic fauna in aquatic sediments. Springer-Verlag, Berlin.

Gotceitas, V., S. Fraser and J.A. Brown (1997) Use of eelgrass beds (*Zostera marina*) by juvenile Atlantic cod (*Gadus morhua*). *Canadian Journal of Fisheries and Aquatic Sciences* 54: 1306–1319.

Grall, J. and M. Glemarec (1997) Biodiversite des fonds de maerl en Bretagne: approche fonctionnelle et impacts anthropogeniques. *Vie Milieu* 47(4): 339–349.

Griffith, C.L. (1976) Guide to the Benthic Marine Amphipods of Southern Africa. Trustees of the South African Museum, Cape Town 106pp.

Gurjanova, E. (1961) Bokoplavy morej SSSR i soprodel'nykh vod (Amphipoda-Gammaridea). *Opred. Po Faune SSSR, Akad. Nauk SSSR* 41: 1–1029.

Harrison, K. and J.P. Ellis (1991) The genera of the Sphaeromatidae (Crustacea: Isopoda): A key and distribution list. *Invertebrate Taxonomy* 5: 915–952.

Hayek, L.A. and M.A. Buzas (1997) Surveying Natural Populations. Columbia University Press, New York, 563pp.

Hemminga, M.A. and C.M. Duarte (2000) Seagrass Ecology. Cambridge University Press, Cambridge 298pp.

Hendler, G., J.E. Miller, D.L. Pawson and P.M. Kier (1995) Sea stars, sea urchins, and allies: Echinoderms of Florida and the Caribbean. Smithsonian Institution Press, Washington.

Hendrickx, M.E. (1995) Camarones. In *Guía FAO para la identificación de especies para los fines de la pesca. Pacífico centro-oriental. Vol. I. Plantas e Invertebrados* *Eds.* W. Fischer, F. Krupp, W. Schneider, C. Sommer, K.E. Carpenter and V.H. Niem FAO, Roma, Italia p. 417–537.

Heywood, V.H. (1995) Global biodiversity assessment. Cambridge University Press, Cambridge.

Higgins, R.P. and H. Thiel (1988) Introduction to the Study of Meiofauna. Smithsonian Institution Press, Washington D.C.

Hillis, D.M., C. Moritz and B.K. Mable (1996) Molecular Systematics 2nd Edition. Sinauer Associates Inc., Sunderland 655pp.

Hirayama, A. (1983–1986) Taxonomic studies on the shallow waters Gammaridean Amphipoda of West Kyushu, Japan. Pub. Seto Marine Biological Laboratory. Vols. 28–31.

Holthuis, L.B. (1980) Shrimps and prawns of the world. FAO Species Catalogue Vol. 1. An annotated catalogue of species of interest to fisheries. FAO Fisheries Synopsis No. 125(1): 1–271.

Holthuis, L.B. (1991) Marine lobsters of the world. FAO species catalogue Vol. 13. An annotated and illustrated catalogue of species of interest to fisheries known to date. FAO Fisheries Synopsis 125(13): 1–292.

Hooper, J. & R.W.M. Van Soest Eds. (2002) *Systema Porifera: A guide to the classification of sponges*, vol. 1 and 2. Kluwer Academic, Plenum, New York.

Hurlbert, S.H. (1984) Pseudoreplication and the design of ecological field experiments. *Col. Monogr.* 54: 187–211.

Hutomo, M. and T. Peristiwady. (1996) Diversity, abundance and diet of fish in the seagrass beds of Lombok Island, Indonesia. *In* Proceedings of an International Workshop, Rottneest Island, Western Australia, 25–29 January 1996 Eds. J. Kuo, R.C. Phillips, D.I. Walker and H. Kirkman, p. 205–212.

Hyman, Libbie (1995) *The Invertebrates, Volume IV, Echinodermata*, McGraw-Hill, New York.

Imbach, M.C. (1967) Gammaridean Amphipoda from the South China Sea. *Naga Report* 4 (1): 39–167.

Irvine, L.M. and Y.M. Chamberlain (1994) Seaweeds of the British Isles Volume 1 Rhodophyta Part 2B Corallinales, Hildenbrandiales. HMSO: London.

Kennelly, S.J. and A.J. Underwood (1984) Underwater microscope sampling of a sublittoral kelp community. *Journal of Experimental Marine Biology and Ecology* 76: 67–78.

Kennelly, S.J. and A.J. Underwood (1985) Sampling of small invertebrates of natural hard substrata in a sublittoral kelp forest. *Journal of Experimental Marine Biology and Ecology* 89: 55–67.

Kensley, B. 1978. Guide to the marine isopods of southern Africa. Trustees of the South African Museum: Cape Town, 173pp.

Kensley, B. and M. Schotte (1989) Guide to the marine isopod crustaceans of the Caribbean. Smithsonian Institution Press: Washington, D.C. and London, 308pp.

Kuo, J. and A.J. McComb (1989) Chapter 2: Seagrass taxonomy, structure and development. *In* *Biology of Seagrasses* Eds. A.W. Larkum, A.J. McComb and S.A. Shepherd. Elsevier, Amsterdam, p. 6–73.

Kuo, J. and C. den Hartog (2001) Chapter 2: Seagrass taxonomy and identification key. *In* *Global Seagrass Research Methods* Eds. F.T. Short and R.G. Coles. Elsevier, Amsterdam, p. 31–58.

- Lamb, A. and P. Edgell (1986) Coastal fishes of the Pacific northwest. Harbour Publishing Co. Maderira, B.C. Canada, 224pp.
- Lambshead, P.J.D. (2003) Marine nematode deep-sea biodiversity — hyperdiverse or hype? *Journal of Biogeography* 30: 475–485.
- Laubits, D.R. (1993) Caprellidea (Crustacea: Amphipoda) towards a new synthesis. *J. Nat. Hist.* 27: 965–976.
- Le Croy, S.E. (1995) Amphipod Crustacea III. Family Colomastigidae. *Mem. Hourglass Cruises* 9(2): 1–139.
- Ledoyer, M. (1982) Crustacés Amphipodes Gammariens Familles des Acanthonotosomatidae á Gammaridae. *Faune de Madagascar* C.N.R.S. Paris 59(1): 1–598.
- Ledoyer, M. (1986) Crustacés Amphipodes Gammariens Familles des Haustoriidae á Vitjazianidae. *Faune de Madagascar* C.N.R.S. Paris 59(2): 599–1112.
- Lincoln, R. J. (1979) British Marine Amphipoda: Gammaridea. *British Mus. Nat. Hist.* London, 658pp.
- Littler, D.S and M.M. Littler (2000) Caribbean Reef Plants. Offshore Graphics, Washington D.C.
- Littler, D.S. and M.M. Littler (2003) South Pacific Reef Plants. Off Shore Graphics, Washington.
- Littler M.M. and D.S. Littler (1985) Nondestructive sampling. In Handbook of phycological methods — field methods: macroalgae Eds. M.M. Littler & D.S. Littler, Cambridge University Press, p. 161–175.
- Loneragan, N.R., R.A. Kenyon, M.D. Haywood and D.J. Staples (1994) Population dynamics of juvenile tiger prawns (*Penaeus esculentus* and *P. semisulcatus*) in seagrass habitats of the western Gulf of Carpentaria, Australia. *Mar. Biol.* 119: 133–143.
- Loreau, M., S. Naeem and P. Inchausti (2002) Biodiversity and ecosystem functioning: synthesis and perspectives. Oxford University Press, New York.
- Love, M. (1996) Probably more than you want to know about the fishes of the Pacific coast. Really Big Press, Santa Barbara, CA, 376pp.
- Lowry, J. and H.E. Stoddard (1997) Amphipoda Crustacea IV. Families Aristiidae, Cyphocaridae, Endeavouridae, Lysianassidae, Scopelocheiridae, Uristidae. *Mem. Hourglass Cruises* 10(1): 1–148.
- Magurran, A.E. (2004) Measuring biological diversity. Blackwell, Malden.
- Mann, K. (1982) Ecology of coastal waters: A systems approach. University of California Press, Berkely, CA 322pp.
- Mares, M.F. (1942) A study of a marine benthic community with special reference to the microorganisms. *J. Mar. Biol. Assoc. U.K.* 25: 517–554.
- Marsh, H., H. Penrose, C. Eros and J. Hugues (2002) Chapter I. Introduction. In Dugong: Status report and action plans for countries and territories. UNEP/DEWA/RS.02–1.

- McArdle, B.H. and M.J. Anderson (2001) Fitting multivariate models to community data: a comment on distance-based redundancy analysis. *Ecology* 82: 290–297.
- McCain, J.C. (1968) The Caprellidea (Crustacea, Amphipoda) of the Western North Atlantic U.S. *Nat. Mus. Bull.* 278: 1–147.
- McManus, J.W. (1993) Managing seagrass fisheries in southeast Asia: An introductory overview. Technical Papers: Study No. 6 Seagrass Resources in Southeast Asia. UNESCO — Jakarta (ROSTSEA), Indonesia p. 83–93.
- Meese, R.J. and P.A. Tomich (1992) Dots on the rocks: a comparison of percent cover estimation methods. *Journal of Experimental Marine Biology and Ecology* 165: 59–73.
- Metsger, D.A. and S.C. Byers (2003) Managing the Modern Herbarium: An Interdisciplinary Approach. The Society for the Preservation of Natural History Collections Reference Manuals, N.Y., 235pp.
- Miller, D.J. and R.N. Lea (1972) Guide to the coastal marine fishes of California. California Department of Fish and Game, *Fish Bulletin* 157: 1–235.
- Minoura, K. and T. Nakamori (1982) Depositional environment of algal balls in the Ryukyu group, Ryukyu Islands, southwestern Japan. *Journal of Geology* 90: 602–609.
- Mollmann, A. and B. Santelices (1997) Flora Marina de Chile Central. Ediciones Universidad Catolica de Chile, Santiago *In Spanish and English*.
- Moore, R. *Ed.* (1966) Treatise on Invertebrate Paleontology, Parts S–U, Geological Society of America, Kansas.
- Mortensen, T. (1928–1951) Monograph of the Echinoidea, Volume I–V. C.A. Reitzel, Copenhagen.
- Orth, R.J., J. van Montfrans, R.N. Lipcius and K.S. Metcalf (1996) Utilization of seagrass habitat by the blue crab, *Callinectes sapidus* Rathbun, in Chesapeake Bay: A review. *In* Proceedings of an International Workshop, Rottnest Island, Western Australia, 25–29 January 1996. *Eds.* J. Kuo, R.C. Phillips, D. I. Walker and H. Kirkman, p. 213–224.
- Ortiz, M. and A. Jimeno (2001) Guía ilustrada para la identificación de las familias y los géneros de los anfípodos del Suborden Gammaridea de la Península Ibérica. *Graellsia* 57(2): 3–93.
- Ortiz, M., A. Martín, I. Wingfield, Y. Díaz and D. Atienza (2004) Anfípodos (Crustacea: Gammaridea) Clave gráfica para la identificación de las familias, géneros y especies marinas y estuarinas del Atlántico Occidental Tropical. UNAM, FES Iztacala, México 162pp.
- Ortiz, M., F. Álvarez and I. Wingfield (2002) Caprellid Amphipods: Illustrated key for the genera and species from the Gulf of Mexico and the Caribbean Sea. UNAM, FES Iztacala 83pp.
- Paine, R.T. (1974) Intertidal community structure: experimental studies on the relationship between a dominant competitor and its principal predator. *Oecologia* 15: 93–120.

- Phillips, R.C. and E.G. Meñez (1988) Seagrasses. Smithsonian Contributions to Marine Science, No. 34. Smithsonian Institution Press, Washington, D.C. 104pp.
- Pettibone, M.H. (1982) Annelida. In Synopsis and Classification of Living Organisms vol. 2. Ed. S.P. Parker, McGraw-Hill, New York, N.Y., p. 1–43.
- Poore, G.C.B. Ed. (2002) Crustacea: Malacostraca: Syncarida, Peracarida: Isopoda, Tanaidacea, Mictacea, Thermosbaenacea, Spelaeogriphacea. In Zoological Catalogue of Australia Vol. 19.2A. Eds. W.W.K. Houston and P.L. Beesley, CSIRO Publishing: Melbourne xii, 429pp.
- Poore, G.C.B. (2001a) Families and genera of Isopoda Anthuridea. *Crustacean Issues* 13: 63–173.
- Poore, G.C.B. (2001b) Isopoda Valvifera: diagnoses and relationships of the families. *Journal of Crustacean Biology* 21: 213–238.
- Poore, G.C.B. and H.M. Lew Ton (1990) The Holognathidae (Crustacea: Isopoda: Valvifera) expanded and redefined on the basis of body-plan. *Invertebrate Taxonomy* 4: 55–80.
- Poore, G.C.B. and H.M. Lew Ton (1993) Idoteidae of Australia and New Zealand (Crustacea: Isopoda: Valvifera). *Invertebrate Taxonomy* 7: 197–278.
- Quinn, G.P. and M.J. Keough (2002) Experimental design and data analysis for biologists. Cambridge University Press, Cambridge.
- Rios, Eliezer (1994) Seashells of Brazil, Fundacao Universidade de Rio Grande, Rio Grande, Brazil.
- Riosmena-Rodriguez, R., W.J. Woelkerling and M.S. Foster (1999) Taxonomic reassessment of rhodolith-forming species of *Lithophyllum* (Corallinales, Rhodophyta) in the Gulf of California, Mexico. *Phycologia* 38(55): 401–417.
- Rivera, M.G., R. Riosmena-Rodriguez and M.S. Foster (2004) Edad y crecimiento de *Lithothamnion muellerii* (Corallinales, Rhodophyta) en el suroeste del Golfo de California, México. *Ciencias Marinas* 30: 235–249.
- Roberts, D.E., S.R. Fitzhenry and S.J. Kennelly (1994) Quantifying subtidal macrobenthic assemblages on hard substrata using a jump camera method. *Journal of Experimental Marine Biology and Ecology* 177: 157–170.
- Roper, C., M. Sweeney and C. Nauen, (1984) Cephalopods of the World, FAO Species Catalogue Vol. 3, FAO Fisheries Synopsis No. 125 Roma, Italy.
- Rose C.L., C.A. Hawks, H.H. Genoways (2002) Storage of Natural History Collections: A Preventive Conservation Approach. The Society for the Preservation of Natural History Collections Reference Manuals NY. 350pp.
- Rose, C.L. and A.R. de Torres (2002) Storage of Natural History Collections: Ideas and Practical Solutions. The Society for the Preservation of Natural History Collections Reference Manuals NY. 260pp.
- Rosenberg, G. (1992) The Encyclopedia of Seashells. Michael Friedman Publishing Group, New York.

Rouse, G.W. and F. Pleijel (2001) Polychaetes. Oxford University Press.

Ruffo, S. *Ed.* (1982) The Amphipoda of the Mediterranean. Part 1. Gammaridea (Acanthonozomatidae to Gammaridae) *Mem. Inst. Ocean., Fond. A. Ier, Prince de Monaco*, 13: 1–364.

Ruffo, S. *Ed.* (1989) The Amphipoda of the Mediterranean. Part 2. Gammaridea (Haustoriidae to Lysianassidae). *Mem. Inst. Ocean., Fond. A. Ier, Prince de Monaco*, 13: 365–576.

Ruffo, S. *Ed.* (1993) The Amphipoda of the Mediterranean. Part 3. Gammaridea (Melphidippidae to Talitridae), Ingolfiellidea, Caprellidea. *Mem. Inst. Ocean., Fond. A. Ier, Prince de Monaco*, 13: 576–813.

Ruffo, S. *Ed.* (1998) The Amphipoda of the Mediterranean. Part 4. Localities. Key to Families. Ecology. Faunistics and Zoogeography. Bibliography. *Mem. Inst. Ocean., Fond. A. Ier, Prince de Monaco*, 13: 814–959.

Rufford, P.J. (1902) Notes on British hydroid zoophytes and other subjects. Burfield & Pennells, Hastings, 149pp.

Ruppert, E. and E. Barnes (1996) Zoología de los invertebrados. Sexta Edición. McGraw-Hill Interamericana. México D. F, México. 1114pp.

Salvat, B. and C. Rives (1975) Coquillages de Polynésie. Les Éditions du Pacifique Papeete, Tahiti.

Schläpfer, F. and B. Schmid (1999) Ecosystem effects of biodiversity: a classification of hypotheses and exploration of empirical results. *Ecol. Appl.* 9, 893–912.

Schroeter, S.C., J.D. Dixon, J. Kastendiek, R.O. Smith, and J.R. Bence (1993) Detecting the ecological effects of environmental impacts: A case study of kelp forest invertebrates. *Ecological Applications* 3(2): 331–350.

Sendall, K. (2003) Phenoxylethanol as a relaxant before fixation in the sea cucumber *Cucumaria miniata* (Echinodermata). *Collection forum* 18: 98–103.

Shirayama, Y., R.P. Higgins and S. Kaku (1993) Microscopic observation of meiofauna from both dorsal and ventral side using HS-slide. *Benthos Res.* 44: 41–44 *In Japanese*.

Shirayama, Y., A.A. Rowden, D.P. Gordon and H. Kawai (2002) Latitudinal biodiversity in coastal macrophyte communities. *In* Biodiversity Research Methods — IBOY in Western Pacific and Asia *Eds.* T. Nakashizuka and N. Stork Kyoto University Press, Kyoto pp. 162–182.

Short, F.T., R.G. Coles and C. Pergent-Martini (2001) Chapter 1: Global seagrass distribution. *In* Global Seagrass Research Methods *Eds.* F.T. Short and R.G. Coles Elsevier Science B.V. Amsterdam, p. 5–30.

Steller, D. L. (2003) Age and growth in rhodolith populations: estimating recovery of the habitat forming corallines *Lithophyllum margaritae* and *Spongites trichotomum*. Chapter 1: Biology Santa Cruz, University of California.

- Steller, D.L. and M.S. Foster (1995) Environmental factors influencing distribution and morphology of rhodoliths in Bahia Concepcion, B.C.S., Mexico. *Journal of Experimental Marine Biology and Ecology* 194: 201–212.
- Steller, D.L., R. Riosmena-Rodriguez, M.S. Foster and C.A. Roberts (2003) Rhodolith bed diversity in the Gulf of California: the importance of rhodolith structure and consequences of disturbance. *Aquatic Conservation-Marine and Freshwater Ecosystems* 13 (Suppl S): S5–S20.
- Stewart-Oaten, A.M. (1986) Environmental impact assessment: 'pseudo-replication' in time? *Ecology* 67(4): 929–940.
- Thomas, J.D. (1997) Systematics, Ecology and Phylogeny of the Anamixidae (Crustacea: Amphipoda). *Rec. Aust. Mus.* 49: 35–98.
- Tsuda, R.T. and I.A. Abbott (1985) Collection, handling, preservation and logistics. In *Handbook of Phycological Methods Ecological Field Methods: Macroalgae* Eds. M.M. Littler and D.S. Littler, Cambridge University Press p. 67–86.
- Underwood, A.J. and Chapman M.G. (1996) Scales of spatial patterns of distribution of intertidal invertebrates. *Oecologia* 107: 212–224.
- Underwood, A.J. (1997) Experiments in Ecology: Their logical design and interpretation using analysis of variance. Cambridge University Press, Cambridge.
- van Soest, R.W.M. (1978) Marine sponges from Curaçao and other Caribbean localities. Part I. Keratosa. *Studies of the fauna of Curaçao and other Caribbean islands* 56(179): 1–94.
- van Soest, R.W.M. (1980) Marine sponges from Curaçao and other Caribbean localities. Part II. Haplosclerida. *Studies of the fauna of Curaçao and other Caribbean islands* 62(191): 1–173.
- van Soest, R.W.M. (1984) Marine sponges from Curaçao and other Caribbean localities. Part III. Poecilosclerida. *Studies of the fauna of Curaçao and other Caribbean islands* 66(199): 1–167.
- van Soest, R.W.M. and J.C. Braekman (1999) Chemosystematics of Porifera: A review. *Memoirs of the Queensland Museum* 44: 569–589.
- Vaught, K.C. (1989) A Classification of the Living Mollusca. American Malacologists Inc., Melbourne, Florida, USA.
- Verheij, E. (1993) Marine plants on the reefs on the Spermonde Archipiélago, SW Sulawesi, Indonesia. Rijksherbarium-Hortus Botanicus, Leiden, Holanda, 320pp.
- Verrill, A.E. (1928) Hawaiian shallow water Anthozoa. *Bulletin Bernice P. Bishop Museum* 49: 1–30.
- Waide, R.B., M.R. Willig, C.F Steiner, G. Mittelbach, L. Gough, S.I. Dodson, G.P. Juday and R. Parmenter (1999) The relationship between productivity and species richness. *Annu. Rev. Ecol. Syst.* 30: 257–300.

- Waycott, M., K. McMahon, J. Mellors, A. Calladine and D. Kleine (2004) A Guide to Tropical Seagrasses of the Indo-West Pacific. James Cook University Press, Townsville, 72pp.
- Wetzer, R., R.C. Brusca and G.D.F. Wilson (1997) The Order Isopoda. *In* Taxonomic atlas of the benthic fauna of the Santa Maria Basin and the Western Santa Barbara Channel. *Eds.* J.A. Blake and P.V. Scott, Santa Barbara Museum of Natural History, Santa Barbara, p. 1–120.
- Whorff, J.S. and L. Griffing (1992) A video recording and analysis system used to sample intertidal communities. *Journal of Experimental Marine Biology and Ecology* 160: 1–12.
- Wicksten, M.K. (1993) A review and a model of decorating behavior in spider crabs (Decapoda, Brachyura, Majidae). *Crustaceana* 64(3): 314–325.
- Wilson, G.D.F. and J. Wägele (1994) Review of the family Janiridae (Crustacea: Isopoda: Asellota). *Invertebrate Taxonomy* 8: 683–747.
- Wilson, R.S., P.A. Hutchings and C.J. Glasby *Eds.* (2003) Polychaetes: An interactive Identification Guide. CSIRO Publishing, Melbourne, Australia.
- Witman, J.D., R.J. Etter and F. Smith (2004) The relationship between regional and local species diversity in marine benthic communities: A global perspective. *Proc. Nat. Acad. Sci.* 101: 15664–15669.
- Woelkerling, W.J. (1996a) Family Sporolithaceae. *In* The Marine Benthic Flora of Southern Australia — Part IIIB. Gracilariales, Rhodymeniales, Corallinales and Bonnemaisoniales. *Ed.* H.B.S. Womersley, Australian Biological Resources Study, Canberra p. 153–158.
- Woelkerling, W.J. (1996b) Subfamily Melobesioideae. *In* The Marine Benthic Flora of Southern Australia — Part IIIB. Gracilariales, Rhodymeniales, Corallinales and Bonnemaisoniales. *Ed.* H.B.S. Womersley, Australian Biological Resources Study, Canberra p. 164–210.
- Woelkerling, W.J. (1996c) Subfamily Choreonematoideae. *In* The Marine Benthic Flora of Southern Australia — Part IIIB. Gracilariales, Rhodymeniales, Corallinales and Bonnemaisoniales. *Ed.* H.B.S. Womersley, Australian Biological Resources Study, Canberra p. 210–214.
- Woelkerling, W.J. (1996d) Subfamily Lithophylloideae. *In* The Marine Benthic Flora of Southern Australia — Part IIIB. Gracilariales, Rhodymeniales, Corallinales and Bonnemaisoniales. *Ed.* H.B.S. Womersley, Australian Biological Resources Study, Canberra p. 2.
- Woelkerling, W.J. (1996e) Subfamily Mastophoroideae (excluding *Hydrolithon*, *Pneophyllum*, *Spongites* & *Neogoniolithon*). *In* The Marine Benthic Flora of Southern Australia — Part IIIB. Gracilariales, Rhodymeniales, Corallinales and Bonnemaisoniales. *Ed.* H.B.S. Womersley, Australian Biological Resources Study, Canberra p. 237–255.
- Woelkerling, W.J. (1988) The coralline red algae: an analysis of the genera and subfamilies of nongeniculate corallinaceae. London, British Museum of Natural History.
- Worm, B. and J.E. Duffy (2003) Biodiversity, productivity and stability in real food webs. *Trends Ecol. Evol.* 18: 628–632.

Yoshida, T., T. Kitayama, J. Tanaka, K. Kogame, M. Baba, S. Kawaguchi, H. Yamamoto and M. Masuda (1998) Marine Algae of Japan. Uchida Rokakuho, Tokyo *In Japanese*.

Zakaria, M.H., B.J. Sidik and O. Hishamuddin (1999) Flowering, fruiting and seedling of *Halophila beccarii* Aschers (Hydrocharitaceae) from Malaysia. *Aquat. Bot.* 65: 199–207.

Zea, S. (1987) Esponjas del Caribe Colombiano. Bogotá, Editorial Catálogo Científico, 286pp.

Section 27 Online References

Cnidaria

Catalog of species, distribution maps, and related information for members of subclass Hexacorallia of class Anthozoa.

Fautin, D. G. (2006) Hexacorallians of the World

<http://geoportal.kgs.ku.edu/Hexacoral/anemone2>

Crustacea

An information retrieval system for crustaceans of the world.

Keable, S. J., G. C. B. Poore and G. D. F. Wilson (2002). Australian Isopoda: Families. Ver. 2

<http://crustacea.net/crustace/isopoda/index.htm>

Pictorial guide to the Crustacea Decapoda of the Eastern Atlantic, the Mediterranean Sea and the adjacent continental waters and a pictorial guide to the Crustacea Amphipoda of northern Europe.

Cédric d'Udekem d'Acoz, Vader, W. Crustikon, Tromsø Museum, University of Tromsø

<http://www.tmu.uit.no/crustikon/Index.htm>

Introduction to the North East Atlantic and Mediterranean Amphipods.

Myers, A., D. McGrath, R. King (2006) Keys to the North East Atlantic and Mediterranean Amphipods

<http://www.amphipods.com/>

World List of Marine, Freshwater and Terrestrial Isopod Crustaceans (a searchable up-to-date list of all the world's species of isopods plus a bibliography).

Kensley, B., M. Schotte, S. Schilling (2006) Smithsonian National Museum of Natural History, Department of Systematic Biology, Invertebrate Zoology

<http://www.nmnh.si.edu/iz/isopod/>

An extended glossary and descriptions of the few Australian asellote isopod species, and a description of Australian isopod diversity.

Wilson George D. F. (Buz) (2006) Isopod Crustacean Research at the Australian Museum

<http://www.personal.usyd.edu.au/~buz/home.html>

Guide to the coastal isopods of California (general biology, keys to species).

Brusca, R. C., V. Coelho and S. Taiti (2001) A Guide to the Coastal Isopods of California. Electronic publication

http://toweb.org/notes/note_id=3004

Colour photographs and discussion of 15 species of Isopods in southern Australia.

Poore, G. C. B. (1996) Isopods of southern Australia (Museum Victoria)

<http://www.mov.vic.gov.au/crust/isopogal.html>

Isopoda glossary with links to details on Cirolanidae isopods.

Brusca, R. (2001) Glossary of Isopoda Technical Terms.

http://tolweb.org/accessory/PEET_Project_on_Cirolanid_Isopods?acc_id=2007

Taxonomic and biological information on species known to occur in Australia.

Government of Australia (2007) The Australian Biological Resources Study (ABRS)

<http://www.deh.gov.au/biodiversity/abrs/online-resources/index.html>

Literature and species lists for Epicaridea.

Shields, J. (2007) Epicaridea: The parasitic Isopods of Crustacea

<http://www.vims.edu/~jeff/isopod.htm>

Echinodermata

Taxonomic resource for the scientific community in which the genera and higher taxa of echinoids can be simply and rapidly identified.

Andrew, B. S. (2004) Echinoid directory illustrated key to families and genera

<http://www.nhm.ac.uk/research-curation/projects/echinoid-directory/index.html>

Guide to Echinodermata. Life history, Systematics and Morphology.

Waggoner, B., B. R. Speer (2007) Introduction to Echinodermata

<http://www.umcp.berkeley.edu/echinodermata/echinodermata.html>

Information concerning meetings and conferences, publications of interest to echinoderm biologists, titles of theses on echinoderms, and research interests, and addresses of echinoderm biologists.

Ahearn, C. *ed.*, The Virtual Echinoderm Newsletter, Smithsonian Museum of Natural History

<http://www.nmnh.si.edu/iz/echinoderm/>

Guide to Echinodermata; Spiny skinned animals, sea urchins, sea stars, and their allies.

Wray, G. A. (1999) Echinodermata; Tree of Life web project

<http://phylogeny.arizona.edu/tree/eukaryotes/animals/echinodermata/echinodermata.html>

Fish

General information and common seagrass species in South Australia (a PDF brochure).

Government of South Australia, Department of Environment and Heritage (2007)

<http://www.environment.sa.gov.au/coasts/pdfs/seagrasses.pdf>

Flora

Guide to the identification of seagrasses in the Great Barrier Reef, Australia (a book in PDF form).

Lanyon, J. (1986) Guide to the identification of seagrasses in the Great Barrier Reef region, Great Barrier Reef Marine Park Authority Special Publication Series, 3

http://www.gbrmpa.gov.au/corp_site/info_services/publications/misc_pub/misc_011/mp_011_full.pdf

Illustrated guide and key to Queensland area Seagrasses (a PDF handout).

Queensland Conservation seagrass and community volunteers (2005) Seagrass Watch Manual for Mapping & Monitoring Seagrass Appendix III

http://www.qccqld.org.au/seagrass/monitoring/sw_monitoring_appendix3.pdf

Species list, pictures, bibliography and general information on seagrasses.

K. W. Bridges, R. C. Phillips *et al.* (2006) Seagrasses

<http://www.botany.hawaii.edu/seagrass/>

Global network of scientists and costal managers committed to research, protection and management of the world's seagrasses.

Philips, R. *et al.* (2006) World Seagrass Association

<http://www.worldseagrass.org>

A database on algae, including terrestrial, marine and freshwater organisms.

Guiry, M. D. *et al.* (2007) AlgaeBase version 4.1. Worldwide electronic publication, National University of Ireland, Galway.

<http://www.algaebase.org/>

A database of seaweed information focused on Africa.

Guiry, M. D. *et al.* (2004) SeaweedsAFRICA.org AlgaeBase, Version 4.1.

<http://www.seaweedafrica.org/>

Checklist of benthic marine flora of Guam, Mariana Islands and Micronesia.

Lobban, C., M. Schefter and T. Schils (2006) Algal* Biodiversity

http://www.uog.edu/classes/botany/474/marine_toc.htm

General Taxonomy

Taxonomic information on plants, animals, fungi and microbes of North America and the world.

The Integrated Taxonomic Information System (ITIS)

<http://www.itis.gov/>

Checklists, maps and information on Italian fauna.

Stoch Fabio (2003) Fauna ItaliaOnline version 2.0, Italian Ministry of the Environment

<http://faunaitalia.it/index.htm>

Gateway to online European resources of the natural world including databases, keys and images.

University of Nottingham (2006)

<http://nature.ac.uk/browse/590.744.html>

Biology teaching resources.

Morin, A., J. Houseman (2007) Digital Resource for Teaching Biology (BIODIDAC)

<http://biodidac.bio.uottawa.ca/>

An searchable megabase with 25472 unique taxa pooled from 20 separate taxonomic databases.

World Biodiversity Database (2007), Expert Center for Taxonomic Identification

<http://nlbif.eti.uva.nl/bis/index.php>

European register of marine species.

Flanders Marine Institute (2006) MarBEF Data System

<http://www.marbef.org/data/erms.php>

A photographic guide to the marine life in Britain and Ireland.

Picton, B., C. Morrow (2002) Encyclopedia of Marine Life of Britain and Ireland

<http://www.habitas.org.uk/marinelife/>

An up-to-date historical cross-referenced classification of life anywhere, anytime based on original authoritative scientific literature.

Brands, S. J. (2005) *Systema Nature 2000*. Amsterdam, The Netherlands
<http://sn2000.taxonomy.nl/>

Phylogenetic trees and systematics.

Tree of Life Web Project (2007) The University of Arizona College of Agriculture and Life Sciences and The University of Arizona Library
<http://tolweb.org/tree/>

Costal and Ocean Plankton Ecology, Production & Observation Database, a global zooplankton and phytoplankton database.

O'Brien, T. (2006) COPEPOD online database US National Marine Fisheries Service, Service & Technology, Marine Ecosystems Division
<http://www.st.nmfs.gov/plankton/search/index.html>

Meiobenthos

The webpage of the International Association of Meiobenthologists, links to their newsletter *Psammonalla* and a list of meiobenthic taxa.

Walters, K. (2006) International Association of Meiobenthologists
<http://www.meiofauna.org/>

Information about nematodes, interactive keys, and resources.

CSIRO Australia (2005) Nematodes (Nematodes or Roundworms)
<http://www.ento.csiro.au/science/nematode.html>

Electronic identification keys to free-living marine nematode diversity.

Plymouth Marine Laboratory, Natural History Museum, London (2006) Darwin Nematode Project
<http://web.pml.ac.uk/nematode/>

A guide to information on gastrotricha, and access to current worm research.

Todaro, M. A. (2007) Gastrotricha Web Portal (2006)
<http://www.gastrotricha.unimore.it/>

Introduction to Nematode research.

Mitreva, M. D. *et al.* (2005) Nematode Net Genome Sequencing Center
<http://www.nematode.net/>

Mollusca

Keys and information on Venezuelan Mollusks (Echinoderms and Crustaceans are also mentioned). *In Spanish*

Bordas H. and G. Griffon (2004) *Le conchiglie dei litorali veneziani*
<http://www.liceofoscarini.it/didattic/conchiglie/>

Searchable Molluscan Key, information and links. *In Japanese and English*

Yamada, M. (2006) Shell Database

<http://www.bigai.ne.jp/index.html>

Macroacology 4.0.2: A database on Western Atlantic Mollusks Species (systematics, biogeography and diversity).

Rosenberg, G. (2005) Malacology 4.0: A database of Western Atlantic marine Mollusca. Electronic publication

<http://data.acnatsci.org/wasp/>

Information on Mediterranean shells with notes on shells from other seas and mega mollusks.

Firm, S. R. L. (2006) Malacology, Electronic publication

http://www.thais.it/conchiglie/default_uk.htm

Online Searchable museum shell collection and information.

Natural History Museum Washington State University

<http://shells.tricity.wsu.edu/>

A bivalve database with images, keys and notes on current research.

Bieler, R. and P. M. Mikkelsen (2005) BIVALVES-Research, Training, Electronic Dissemination of Data

<http://peet.fnmh.org/>

Images of seashells from the Alboran Sea and North West Africa, Morocco to Guinea including the Canary Island.

Belloq, M., J. M. Hernández, Otero and F. Gubbioli (2006) Alboran Shells

<http://www.alboranshells.com/>

A database-driven web site on all living cephalopods (octopus, squid, cuttlefish and nautilus), taxonomy, images and information

Wood, J. B. and C. Day (2006) CephBase Dalhousie University

<http://www.cephbase.utmb.edu/>

Physical Oceanography

Information on physical oceanography.

Stewart, R. H. (2005) Introduction to Physical Oceanography

http://oceanworld.tamu.edu/ocean410/ocng410_text_book.html

Satellite maps.

Landsat, ASTER, MODIS and AVHRR

<http://glcfapp.umiacs.umd.edu:8080/esdi/index.jsp>

Global sea level data.

Global Sea Level Observing System (GLOSS)

<http://www.pol.ac.uk/psmsl/programmes/gloss.info.html>

Global climate and (some) marine data.

Center of the U.S. Department of Commerce

<http://www.ncdc.noaa.gov/oa/ncdc.htm>

Global Weather forecast.

World Meteorological Organization WMO

<http://www.worldweather.org/>

Ocean Colour data.

SeaWiFS, MODIS

<http://oceancolor.gsfc.nasa.gov/>

Ocean Wind data.

NASA Jet Propulsion Laboratory

<http://podaac.jpl.nasa.gov/>

Polychaeta

Online identification key to Polychetes. *In French*

Narendra Jussinen (2006) NEREIS Electronic publication

<http://www.ima.uco.fr/etudiants/projets/nereis/index.html>

A guide to information on annelids, and current worm research and researchers.

Read, G. B. (2006) Annelida Resources Electronic publication

<http://www.annelida.net/>

Online bibliography containing 17500 references from the last 300 years on polychaete annelids.

Ward, L. and K. Fauchald (1997) Polychaete Bibliography, 2nd Edition

http://134.60.85.50:591/PolyDB/PolyDBN_su.html

Taxonomic information about polychaetes by the Darwin Initiative project.

Paterson, G., Kendall, M., Aryuthaka, C. (2005) Taxonomic information across the internet; Natural History Museum of London

<http://www.nhm.ac.uk/research-curation/projects/taxinfo/index2.html>

An interactive key and information system for polychaete families and higher taxa.

Glasby, C. J. and K. Fauchald (2003) Polikey Version 2: 5

<http://www.deh.gov.au/biodiversity/abrs/online-resources/polikey/index.html>

Online key to species of the Family Eunicidae found in Italy.

Ponti, M. (2006) Euniidae of Italy

http://www.ecology.unibo.it/Polychaete_keys/Eunicidae/index.htm

Porifera

Guide to Poriferans (Sponges). Life history, Systematics and Morphology.

Collins, A. G., B. Waggoner (2006) Introduction to Porifera Electronic publication

<http://www.ucmp.berkeley.edu/porifera/porifera.html>

A global list of extant sponges — a searchable preliminary database of all living and described sponges.

van Soest, R., N. Boury-Esnault, D. Janussen and J. Hooper (2005) World Porifera database

<http://www.vliz.be/vmcdedata/porifera/>

A guide to classification and identification of Sponge Porifera.

Hooper, J. N. A. *et al.* (2000) 'SPONGUIDE' Guide to sponge collection and identification (Ver. Aug. 2000)

<http://www.qmusem.qld.gov.au/organisation/sections/SessileMarineInvertebrates/index.asp>

Key to Porifera focusing on the class Demospongiae taken from Kozloff, 1987, 1996.

Cowles Dave (2005) Phylum Porifera, University of Washington Press

http://www.rosario.wvc.edu/inverts/Porifera/Porifera_Key.html

Field Guide to Sponges from Antarctica.

Conlan, K., P. Cziko, J. Mastro and N. Wu (1998). Underwater Field Guide to Ross Island & McMurdo Sound, Antarctica; Canadian Museum of Nature

<http://scilib.ucsd.edu/sio/nsf/fguide/porifera.html>

Species lists of Porifera found globally. The Porifera collection of the ZMA, database, catalogue and 3D images.

van Soest, R. The Porifera collection of the Zoologisch Museum, Amsterdam

<http://www.science.uva.nl/zma/databases.cfm> (Choose: Porifera)

Tunicata

Up-to-date listings of recent publications on ascidians worldwide, and ascidian abstracts from recent meetings and links.

Lambert, C. C. and G. Lambert (2006) Ascidian Home Page for the United States

<http://depts.washington.edu/ascidian/>

Section 28 Contributors

Todd W. Anderson is an Associate Professor in the Department of Biology, San Diego State University, 5500 Campanile Drive, San Diego, California, 92182-4614, USA. He conducts research on the ecology of fishes, particularly the population and community ecology of temperate and tropical reef fishes. He can be contacted by phone (619-594-0995) or e-mail (todda@sunstroke.sdsu.edu). Information about Dr. Anderson's research, graduate students, publications, presentations, and current projects can be found at his "Fish Ecology Lab" website (<http://www-rohan.sdsu.edu/~kelpbass>). *Contribution: Protocols for sampling and monitoring nearshore fishes*

Chittima Aryuthaka is the director of the Ranong Coastal Resource Research Station, Kasetsart University, Ranong Province and is a regional expert on seagrasses under the UNEP/GEF Project: Reversing Environmental Degradation Trends in the South China Sea and Gulf of Thailand. Primarily focused on ecological processes in seagrass beds her interests include ecology and taxonomy of meiobenthos and using the Internet as a tool to increase interest in taxonomy at large. *Contribution: Introduction to seagrass bed ecology and the seagrass identification guide*

Lisandro Benedetti-Cecchi is an Associate Professor in Ecology at the University of Pisa. His research focuses primarily on the ecology of assemblages of algae and invertebrates in intertidal and subtidal rocky seashores. Key-research topics and expertise include: design and analysis of ecological experiments; analysis of the roles of species' interactions and climate change in determining patterns of biodiversity; experimental evaluation of consistency and variation of ecological processes at multiple scales in space and time; development of experimental designs to separate the effects of intensity and temporal variance of ecological processes; optimisation of environmental sampling designs for impact assessment; species invasion and ecology of Marine Protected Areas. He is also the

leader for the NaGISA European Seas region. Web Site: <http://www.discat.unipi.it/BiolMar/people/LBC/LBC.htm> *Contribution: Sampling design*

Juan M. Díaz is a malacologist and marine ecologist. He is the author of more than 60 contributions in scientific journals, most of them dealing with Caribbean molluscs (taxonomy and biogeography), coral reefs and seagrass beds. He has also written several books that describe the distribution and features of marine environments in Colombia and an exhaustive field guide of molluscs of the Colombian Caribbean. He was the leader of the Marine Biodiversity Program at The Instituto de Investigaciones Marinas y Costeras -INVEMAR- in Santa Marta, Colombia, for 18 years. At present, he is Associate Researcher of the Humboldt Institute in Bogotá and scientific advisor of The Nature Conservancy for conservation planning of the Tropical Eastern Pacific region. His telephone number is 57-1-6278858, and his e-mail address is jmdiaz@humboldt.org.co. *Contribution: Mollusca identification guide*

Maria Cristina Diaz specializes in the taxonomy of intertidal and tropical sponges but has a special interest in biodiversity and community structure of the Caribbean. She has been fascinated with sea life since her very first dive and has been working on sponges since focusing on the coral reef of Los Roques National Park for her undergraduate research project. When not being followed by film crews (as she was for the Shape of life project <http://www.pbs.org/kcet/shapeoflife/>) or getting species named after her (*Halicnemia diazae* Desqueyroux-Faúndez & Van Soest, 1997), she can be reached at crisdiaz@ix.netcom.com or taxochica@hotmail.com or found at the Museo Marino de Margarita in Venezuela at the Smithsonian Institute, National Museum of Natural History. *Contribution: Sponge identification guide*

Daphne G. Fautin is a student of sea anemones, specializing in their natural history (reproduction and symbiosis) and evolution (their relationship to scleractinian corals). She is a Professor in the Department of Ecology and Evolutionary Biology at the University of Kansas, USA, and a Curator in the University of Kansas Natural History Museum and Biodiversity Research Center. Her office is in Haworth Hall, University of Kansas, 1200 Sunnyside Avenue, Lawrence, Kansas 66045-7534 USA. Her telephone number is 1-785-864-3062, and her e-mail address is fautin@ku.edu. Her CV, a list of her students, and other related information is on her website, www.nhm.ku.edu/~inverts. The website has a link to her taxonomic and biogeographic database of "Hexacorallians of the World" (reef-forming corals, black corals, tube anemones, sea anemones, etc.): <http://geoportal.kgs.ku.edu/Hexacoral/anemone2>. *Contribution: Cnidarian identification guide*

Michael S. Foster is a Professor Emeritus at Moss Landing Marine Laboratories. He obtained his PhD in biology (1972) from the University of California, Santa Barbara and is a Fellow of the California Academy of Sciences and a Fulbright Scholar. His research interests center on the ecology of subtidal and intertidal reefs, particularly algal populations and assemblages in kelp forests, rhodolith beds and rocky shores, and have resulted in 70 peer-reviewed professional papers, 4 books and numerous reports. He is also interested in anthropogenic impacts to these habitats, and

has worked and published on the impacts of oil spills (1969 Santa Barbara and Exxon Valdez), as well as on the effects of coastal power plant cooling systems on the marine environment. Much of this work has focused on assessing recovery from such impacts, including the application of innovative analytical and statistical analyses. *Contribution: Sampling and monitoring rhodolith beds*

Michel E. Hendrickx leads the Universidad Nacional Autonoma de Mexico benthic invertebrate laboratory, which is oriented towards investigating marine and brackish water invertebrates of the Mexican Pacific. Beyond taxonomy his interests carry over to ecology, biogeography and the anthropogenic effect on natural systems. He has updated several invertebrate inventories of the southern regions (Gulf of California, Mexican Pacific & Tropical Pacific) and carried out numerous revisions of taxa all while continuing to compile a considerable invertebrate specimen collection. He can be reached at Unidad Académica Mazatlán, ICML, UNAM, PO Box 811 Mazatlán, Sinaloa 82000, Mexico. E-mail: michel@ola.icmyl.unam.mx. *Contribution: Decapod identification guide*

Eric C. Henry is a phycologist with a special interest in the systematics and biology of brown algae and culture studies. He is presently an independent consultant providing expertise on algal problems, especially mass culture growth systems. He holds an Affiliate appointment at Oregon State University, and can be contacted at henryc@science.oregonstate.edu or eric@reedmariculture.com. *Contribution: Macroalgae identification guide*

Katrin Iken's research is centered on trophic interactions between organisms. She is generally interested in shallow water community diversity, dynamics and ecology, especially in both Polar Regions, and in deep-sea communities. Most of her shallow water work uses scientific diving as a research tool. She is the NaGISA regional manager of the Polar Seas Region. Details of her current research, teaching schedule and supervising roles can be seen at her page on the University of Alaska, Fairbanks website: <http://www.sfos.uaf.edu/directory/faculty/iken/>. *Contribution: Editor*

Testuya Kato currently at Kyoto University Aquarium is a polychaeta taxonomist with a special interest in the family Phyllodocidae. He has accumulated quite a few frequent flyer miles in the quest of his research including trips around Japan during his doctoral work at Hokkaido University, through Europe specifically Paris to work at the National Museum of Natural History and throughout Asia as a taxonomy workshop leader and field researcher. *Contribution: Polychaete identification guide, Protocols for rocky shores and seagrass beds.*

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Contribution: Sampling and monitoring rhodolith beds

Section 29 Index

- Abdomen 48, 60–65, 75–78
Ambulacrum 43
Antenna 48–50, 60–62, 65, 74, 76–77
Appendage 47–49, 59, 61, 73, 75
Area 7–8, 13–14, 17, 25, 35–36, 44–45, 58, 60, 86, 90–91, 95
Article 77
Ascocyst 35
Axial 50, 58, 96
- Beak 40–41
Benthic 3–4, 15, 22, 24, 47, 60–62, 69, 73–74, 80, 93–94, 96
Body whorl 58
Branchia 48, 57, 59–62, 78
Branchial cavity/chamber 56–57, 59, 69–70, 72, 74, 78, 95
- Calyx 46
Cancellate 58
Carapace 59–62, 64–65
Carpospore 34
Carposporophyte 36
Cephalothorax 59
Chaetae 47–51
- Chela 59, 61–64, 71, 75
Choanocyte 70, 72
Coenocyte 34
Columella 58
Concentric 46
Continental shelf 60, 85
Core/Corer 8, 25–29, 38
- Depth contour (Transect) 17–19, 26, 28, 91–92, 94–95
- Endopod 77
Exopod 77–78
- Flagellum 74, 77
- Gamete 34, 51
Gametophyte 35
Gnathopod 73–74
- Head 47–49, 56–57, 59, 73–77
Hinge 57–58
Hydropore 42
- Lip 51, 58

- Madreporite 42–44
 Mandible 51, 76–78
 Marsupium 74, 76
 Maxilla (1 & 2) 51, 76
 Maxilliped 74–77
 Medusa 52–54
 Meristem 35–36
 Metamorphosis 42–43
 Mutable collagenous tissue 42–43

 Nematocyst 53–54
 Neuropodia/Neuropodium 48
 Notopodia/Notopodium 48

 Operculum 14, 48, 78
 Oscule(a) 69–71

 Palp 48, 50–51, 74–75
 Parapodia/Parapodium 47–50
 Pedicellariae 43–45
 Peduncle (protopod or sympod)
 40, 71, 74, 77
 Pelagic 24, 43, 52, 54, 59–62
 Pentamery 42
 Percent cover 18, 94–96
 Pereon 73–74, 76–77
 Pereonite 74, 77
 Pereopod 59, 61–62, 73–74,
 76–78
 Peristomium 48, 51
 Pleon 73–74, 77–78
 Pleonite 74, 77–78
 Pleopod(s) 73–76, 78
 Pleotelson 76–78
 Plication 3, 9, 45
 Polyp 33, 52–55, 57–58
 Proboscis 48–50
 Prostomium 47–48, 51
 Protoconch 58
 Pygidium 48–49

 Quadrat 8, 18–20, 27
 Quadrat set 27

 Radial 42, 44, 52
 Radial canal 44
 Ray 71
 Reticulate 71
 Rhodophyceae 33, 93
 Rostrum 60–62, 64–65, 74
 Rudiment 42–43

 Seta 76–77
 Site 9, 17–19, 25–28, 86, 94–95,
 97
 Slate 91–92, 94
 Spicule 57, 69–72
 Spiral 58
 Spire 58
 Sporophyte 35–36
 Stereom 42–43, 45, 49–50, 74,
 76, 81
 Stone canal 42
 Subchelate 64

 Taxon (pl. taxa) 8, 21, 29, 35–36,
 42, 44–45, 61, 72, 80, 82
 Tetraspore 34
 Thallus 35–36, 60
 Transect 17–19, 25–26, 28, 90–92,
 94–95
 Tube foot (feet or podia) 42–46

 Ulvophyceae 33
 Umbilicus 58
 Umbo 57
 Uropod 48, 73–74, 76–78

 Varix 58

 Water ring 42
 Water vascular system 42–43
 Whorl 58

 Zoospore 33–34



Natural Geography In Shore Areas (NaGISA), a Census of Marine Life (CoML) project, is aimed at assessing nearshore biodiversity on a global scale. NaGISA promotes simple standardized protocols and focuses on widespread nearshore habitats. This text introduces the reader to the basic concepts of biodiversity-sampling and coastal habitat ecology while outlining NaGISA's standardized field methods for macroalgal and seagrass communities. Written by premier coastal ecologists and taxonomists it has been organized into short, easy to read sections, making it ideal as a text for field courses, a manual for coastal managers or as a reference guide for those attempting to inventory species or monitor their local shoreline.



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